Papers and Proceedings of

The Royal Society *of* Tasmania

Volume 153
December 2019

MUSEUMS VICTORIA
1 2 FEB 2020

Papers and Proceedings of

The Royal Society *of* Tasmania

Edited by John K. Volkman and M. Davies
and published by the Society

Volume 153
December, 2019

Published by
The Royal Society of Tasmania
GPO Box 1166
Hobart, Tasmania, Australia 7000

www.rst.org.au

14 December 2019

ISSN 0080-4703

Cover illustration: Painted apple moth (*Orgyia anartoides*).

Print Tasmania

PAPERS AND PROCEEDINGS OF THE ROYAL SOCIETY OF TASMANIA

VOLUME 153

Contents

page

Volkman, J.K. — The Royal Society of Tasmania commemorates 175 years of service to Tasmania 1

Baker, M.L., Grove, S., de Salas, M.F., Byrne, C., Cave, L., Bonham, K., Moore, K. & Kantvilas, G. — Tasmanian Museum and Art Gallery's Expedition of Discovery I – The flora and fauna of *Wind Song*, Little Swanport, Tasmania 5

Magni, P.A., North, J.D., Zwerver, M., Dadour, I.R. — Insect succession pattern on decomposing pig carcasses in Tasmania: a summer study 31

Wapstra, M. — Taxonomic and conservation status of *Dockrillia striolata* (Rchb.f.) Rauschert (Orchidaceae) in Tasmania 39

Nichols, E.C. & Kirkpatrick, J.B. — Patterns and causes of marsupial paths in subalpine Tasmania 53

Aalders, J., McQuillan, P. & Prahalad, V. — Vegetation communities and edaphic relationships along a typical coastal saltmarsh to woodland gradient in eastern Tasmania 61

Tasmania
Explore the possibilities

UNIVERSITY of TASMANIA

Publication of this volume was generously supported by the Government of Tasmania and the University of Tasmania.

THE ROYAL SOCIETY OF TASMANIA

Council and Office Bearers from March 2019 to March 2020

Patron
Her Excellency Professor the Honourable Kate Warner AC, Governor of Tasmania

President
Prof. Ross Large AO

Vice President
Mrs Mary Koolhof

Immediate Past President
Prof. Matt King

Honorary Secretary
Mr David Wilson

Honorary Treasurer
Mr Peter Meyer

Councillors
Dr Robert Johnson
Dr Greg Lehman
Dr Angela Ryan
Mrs Roxanne Steenbergen
Dr John Thorne AM
Dr Adele Wilson

Early Career Researcher
Dr Taryn Noble

Student Council Member
Mr Austen Hawkins

Co-opted Council Members
Dr Anita Hansen (co-opted to October 2019)
Ms Jeannie-Marie Leroi (Deputy Publicity Officer)

Honorary Editor
Dr Margaret Davies OAM
Dr John Volkman (from April 2019)

Honorary Librarian
Ms Juliet Beale

Honorary Publicity Officer
Dr Adele Wilson

Honorary Membership Secretary
Mrs Roxanne Steenbergen

Representative of the Tasmanian Museum and Art Gallery
Ms Janet Carding

Representatives of the Northern Chapter
Ms Chel Bardell
Mr David Morris

THE ROYAL SOCIETY OF TASMANIA COMMEMORATES 175 YEARS OF SERVICE TO TASMANIA

by John K. Volkman

(with three plates)

Volkman, J.K. 2019. (14:xii). The Royal Society of Tasmania commemorates 175 years of service to Tasmania. *Papers and Proceedings of the Royal Society of Tasmania* 153: 1–4. ISSN 0080-4703. Honorary Editor, The Royal Society of Tasmania, GPO Box 1166, Hobart, Tasmania 7000, Australia (JKV). E-mail: johnkvolkman@gmail.com.

In 2018, The Royal Society of Tasmania commenced its celebration of 175 years of service to the people of Tasmania. As the culmination of these celebrations a special plaque was unveiled at the Argyle Street entrance to the Tasmanian Museum and Art Gallery on 14 October 2019 by the Society's Patron Her Excellency Professor the Honourable Kate Warner AC, Governor of Tasmania together with Professor Ross Large AO, President of The Royal Society of Tasmania and Ms Janet Carding, Director of the Tasmanian Museum and Art Gallery.
Key Words: Aboriginal people, Royal Society of Tasmania, Tasmanian Museum and Art Gallery, anniversary plaque.

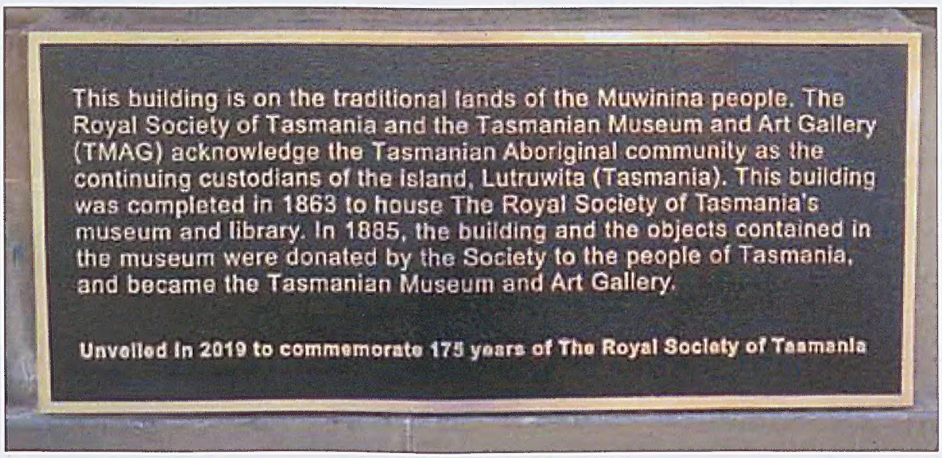

This building is on the traditional lands of the Muwinina people. The Royal Society of Tasmania and the Tasmanian Museum and Art Gallery (TMAG) acknowledge the Tasmanian Aboriginal community as the continuing custodians of the island, Lutruwita (Tasmania). This building was completed in 1863 to house The Royal Society of Tasmania's museum and library. In 1885, the building and the objects contained in the museum were donated by the Society to the people of Tasmania, and became the Tasmanian Museum and Art Gallery.
Unveiled in 2019 to commemorate 175 years of The Royal Society of Tasmania

PLATE 1 — The plaque installed next to the Argyle Street entrance to TMAG (B. Pryor).

INTRODUCTION

The final official event for the year-long commemoration of the 175th Anniversary of The Royal Society of Tasmania took place on Monday, 14 October 2019 at the Tasmanian Museum and Art Gallery (TMAG) in Hobart. It was hosted by the Director of the Tasmanian Museum and Art Gallery, Ms Janet Carding, who gave the welcome address and introduced the speakers, Professor Ross Large AO, President of The Royal Society of Tasmania and Her Excellency Professor the Honourable Kate Warner AC, Governor of Tasmania and Patron of the Society. Their speeches are given below. Following the unveiling (pls 1–3) a presentation was made to Dr Anita Hansen in recognition of her sterling work as Convener of the 175th Anniversary Committee. A full listing of anniversary events compiled by Dr Hansen is provided following the text of the speeches.

SPEECH BY THE PRESIDENT OF THE ROYAL SOCIETY OF TASMANIA, PROFESSOR ROSS LARGE AO

Thank you, Janet.

The plaque that the Governor, our Patron, is about to unveil represents 175 years of continuous service of The Royal Society of Tasmania. The Royal Society of Tasmania (RST) has a long and proud history and was instrumental in the development of the colony of Tasmania especially in its first 100 years. This includes establishing the Royal Tasmanian Botanical Gardens (RTBG), the Tasmanian Museum and Art Gallery (TMAG), the Salmon Ponds in the Derwent Valley, the first meteorological stations, and was involved in other key issues such as the exploration of the Tasmanian landscape, promotion of the first areas for National Parks and experimentation and identification of suitable industries for Tasmania. Today the RST continues as a scientific (and historical) society with the mission of "The advancement of knowledge".

The 175th Anniversary year has been an excellent year for The Royal Society of Tasmania. Dr Anita Hansen (Convener) and the 175th Anniversary Committee have worked tirelessly to produce an outstanding array of events for our 175th celebrations.

A list of the major public events includes:

- The *Dinosaur rEvolution* exhibition, held jointly with TMAG which had over 60,000 visitors over 5½ months.
- The *Dinosaur Picnic in the Park*, held jointly with the Royal Tasmania Botanical Gardens which attracted over 2,000 Tasmanian families.
- The publication of two magnificent books based on the rare book archives of RST: *Mapping Van Diemen's Land and the Great Beyond* edited by Lynn Davies, Margaret Davies and Warren Boyles; and secondly, *Poles Apart: Fascination, Fame and Folly* edited by Anita Hansen and Brita Hansen.
- An anniversary play, *The Laughing Gas Cure*, written by Stella Kent was presented by our Northern Branch with great success.
- Two art exhibitions, one in February jointly with the Art Society of Tasmania featuring the RST art of Louisa Anne Meredith and another by TMAG in March featuring the RST artworks of Simpkinson de Wesselow.
- Finally, a scientific symposium: *Dinosaurs and Evolution of Life* featuring talks by dinosaur experts and palaeontologists from around Australia in March this year.

During this 175th year, the Society also formed an Aboriginal Engagement Committee with co-chairs Dr Gregory Lehman and Professor Matt King to investigate past actions by the RST in relation to Tasmanian Aboriginal people and develop a strategy for reconciliation.

I am proud and immensely privileged to be the President of a scientific society with such a significant historical role in the development of Tasmania, but also a society made up of members — volunteers who all want to give back something to their State by way of encouraging younger Tasmanians through scholarships, lectures, symposiums, dinosaur exhibitions, art exhibitions etc. It all comes back to the advancement of knowledge for all ages. My proudest moment this 175th year was seeing all the young children having so much fun at our dinosaur exhibition and the dinosaur picnic in the gardens. These were collaborations with TMAG and RTBG, the two organisations that the RST founded and ran for 40 years from 1843 to 1885. I have also been incredibly proud of our RST Council and associated members who have worked tirelessly over our 175th year, to bring such a varied and exciting program of events to the general public of Tasmania.

PLATE 2 — Professor Ross Large AO, President of The Royal Society of Tasmania, Ms Janet Carding, Director of the Tasmania Museum and Art Gallery and Her Excellency Professor the Honourable Kate Warner AC, Governor of Tasmania and Patron of the Society (B. Pryor).

SPEECH BY HER EXCELLENCY PROFESSOR THE HONOURABLE KATE WARNER AC GOVERNOR OF TASMANIA

Thank you, Janet.

As we have heard from Ross, The Royal Society of Tasmania's anniversary year has been well and truly celebrated and commemorated as it should be, given the Society's great age in the Australasian context, and its status as one of the oldest such societies outside the UK.

Furthermore, the Society, as the progenitor of both the Royal Tasmanian Botanical Gardens and the Tasmanian Museum and Art Gallery, lays just claim to being a pre-eminent Australian scientific, cultural and intellectual body. This is underscored through the Society's close association with the University of Tasmania, through its Library being housed at the University; while the Northern Branch is based at Launceston's Queen Victoria Museum and Art Gallery. Further proof of your status as a repository of science and culture lies in the *Papers and Proceedings of the Royal Society of Tasmania*, an amazing historical collection of scientific papers from the mid-nineteenth century to today.

And the Society's relationship with the Office of the Governor is particularly strong and enduring – albeit with a mildly controversial initiating element. Prior to your founding as the Botanical and Horticultural Society of Van Diemen's Land in 1843, by Lieutenant-Governor Sir John Eardley-Wilmot, his predecessor Sir John Franklin had established The Tasmanian Society in 1841, which published *The Tasmanian Journal of Natural Science, Agriculture [and] Statistics*. Immediately following the departure of Franklin, a split occurred in that body, leading to the creation, at the behest of Eardley-Wilmot, of a new society on Saturday 14 October 1843. And the following year Eardley-Wilmot secured Queen Victoria's patronage and you became "The Royal Society of Van Diemen's Land for Horticulture, Botany and the Advancement of Science".

Meetings of the Royal Society were held in the Harrington Street Museum from 1852; and in 1861 Governor Sir Henry Fox Young, as President, laid the foundation stone of The Royal Society of Tasmania's museum and library.

Volume 2 of the *Papers and Proceedings of the Royal Society of Tasmania* contains two papers from W.T. Denison, who was the last Lieutenant Governor of Van Diemen's Land before it was granted self-government and became Tasmania. The first paper was titled, "On various modes of planting the potato", and the second dealt with "manures most suitable for turnips". A later Governor, Frederick Weld, was co-author of a number of papers dealing with the introduction of salmon, related no doubt to the establishment of the Salmon Ponds, which the Royal Society was responsible for.

I will quote now the words of the plaque that is about to be unveiled:

"This building is on the traditional lands of the Muwinina people. The Royal Society of Tasmania and the Tasmanian

PLATE 3 — Dr Anita Hansen and Professor Ross Large AO at the plaque unveiling (B. Pryor).

Museum and Art Gallery acknowledge the Tasmanian Aboriginal community as the continuing custodians of the island, Lutruwita (Tasmania). This building was completed in 1863 to house The Royal Society of Tasmania's museum and library. In 1885, the building and the objects contained in the museum were donated by the Society to the people of Tasmania and became the Tasmanian Museum and Art Gallery."

While it took some years for the Society to find its true home, patronage has been central to the Society's identity virtually since its founding, with Queens and Kings as Patrons and Governors as Presidents – until in 2003 the Governor of Tasmania became Patron.

I am honoured to continue in that role, and it is a great privilege for me to be able to demonstrate that by now unveiling this plaque, as a permanent marker of the great achievements past and to come of The Royal Society of Tasmania.

LIST OF EVENTS CELEBRATING THE 175TH ANNIVERSARY

This list was compiled by Dr Anita Hansen as Convener of the 175th Anniversary Committee which organised all of the commemorative events.

19 July 2018: *Mapping Van Diemen's Land and the Great Beyond* – Book launch at the Stanley Burbury Theatre, University of Tasmania, Sandy Bay campus.

30 August 2018: Royal Society of Tasmania medal presentation and lecture at Government House, Hobart.

30 September 2018: *The Laughing Gas Cure* – Play reading at the Queen Victoria Museum and Art Gallery, Inveresk, Launceston.

14 October 2018: *Mapping Van Diemen's Land and the Great Beyond* – Book launch at the Queen Victoria Museum and Art Gallery, Inveresk, Launceston.

18 October 2018; Reception to mark The Royal Society of Tasmania's 175th Anniversary at Government House, Hobart.

6 November 2018: *Poles Apart: Fascination, Fame and Folly* – Book launch at the Tasmanian Museum and Art Gallery, Dunn Place, Hobart.

10 November 2018: *Dinosaur Picnic* at the Royal Tasmanian Botanical Gardens, Queens Domain, Hobart.

13 November 2018: *Poles Apart: Fascination, Fame and Folly* – Exhibition at the Morris Miller Library, University of Tasmania, Sandy Bay campus.

21 November 2018: Book stall at Fullers Book Shop Christmas sale in Hobart.

25 November 2018: *Poles Apart: Fascination, Fame and Folly* – Book launch at the Queen Victoria Museum and Art Gallery, Inveresk, Launceston.

6 December 2018: Launch of *DINOSAUR rEVOLUTION* in Hobart.

11 December 2018: Royal Society of Tasmania members sneak peek at *DINOSAUR rEVOLUTION.*

7 December 2018 to 5 May 2019: *DINOSAUR rEVOLUTION* at the Tasmanian Museum and Art Gallery, Dunn Place, Hobart.

2–24 February 2019: *Louisa Anne Meredith: A Remarkable Woman* art exhibition at the Lady Franklin Gallery, Ancanthe Park, Lenah Valley, Hobart.

23–24 March 2019: *Dinosaur Symposium* at the Stanley Burbury Theatre, University of Tasmania, Sandy Bay campus.

21 March to 11 June 2019: *Simpkinson De Wesselow* art exhibition at the Tasmanian Museum and Art Gallery, Dunn Place, Hobart.

16 August 2019: Sponsorship of a talk at Beaker Street in Hobart.

3 October 2019 to 3 March 2020: *Louisa Anne Meredith: A Remarkable Woman* art exhibition at the East Coast Heritage Museum in Swansea.

14 October 2019: Unveiling of the 175th anniversary plaque on the Tasmanian Museum and Art Gallery building in Hobart.

ACKNOWLEDGEMENTS

I am grateful to Professor Ross Large AO and Her Excellency Professor Kate Warner AC for making their speeches available for publication, to Bernard Pryor for photographs and to Dr Anita Hansen for the compilation of events organised by the 175th Anniversary Committee.

TASMANIAN MUSEUM AND ART GALLERY'S EXPEDITION OF DISCOVERY I – THE FLORA AND FAUNA OF *WIND SONG*, LITTLE SWANPORT, TASMANIA

by Matthew Baker, Simon Grove, Miguel de Salas, Catherine Byrne, Lyn Cave, Kevin Bonham, Kirrily Moore and Gintaras Kantvilas

(with 15 plates, two tables and an appendix)

Baker, M.L., Grove, S., de Salas, M.F., Byrne, C., Cave, L., Bonham, K., Moore, K. & Kantvilas, G. 2019 (14:xii): Tasmanian Museum and Art Gallery's Expedition of Discovery I – The flora and fauna of *Wind Song*, Little Swanport, Tasmania. *Papers and Proceedings of the Royal Society of Tasmania* 153: 5–30. ISSN 0080–4703. Tasmanian Museum and Art Gallery, GPO Box 1164, Hobart, Tasmania 7001, Australia (MLB*, SG, MFS, CB, LC, KB, KM, GK). *Author for correspondence. Email: matthew.baker@tmag.tas.gov.au

A flora and fauna survey was conducted at the east coast Tasmanian property *Wind Song* in 2017 as part of the Tasmanian Museum and Art Gallery's ongoing research, collection-building and nature-discovery program. The survey recorded 885 taxa, primarily from the targeted groups of vascular plants, bryophytes, lichens, butterflies, moths, beetles, snails and slugs. Several of the taxa recorded, chiefly lichens and invertebrates, are new to science or new records for Tasmania. The survey provides a benchmark for further work and serves as an indicator of the biodiversity of a former farming property on Tasmania's east coast.

Key Words: species discovery, biodiversity, Tasmania, lichens, multidisciplinary survey.

INTRODUCTION

In 2017, the Tasmanian Museum and Art Gallery (TMAG) launched its *Expeditions of Discovery* program with the aims of: building TMAG's collections of flora and fauna from under-sampled parts of Tasmania; documenting the species of plants and animals recorded; discovering new or hitherto overlooked species; and highlighting the role that baseline species-discovery research plays in understanding and managing Tasmania's remarkable biota. This initiative for TMAG was inspired by the Australian Government's *Bush Blitz* species discovery program. TMAG has been a part of the *Bush Blitz* program over many years, collaborating in surveys in remote Tasmanian locations such as the Tasmanian Wilderness World Heritage Area, Flinders Island and the Tarkine region (Bauer *et al.* 2010, Commonwealth of Australia 2012, 2014a, 2014b, 2016, 2017a, 2017b, 2017c, Byrne 2017). Together these surveys recognised some 335 putative new species. In Tasmania, multidisciplinary flora and fauna studies, such as the Lower Gordon River Scientific Surveys conducted in the late 1970s (for example Bratt 1978, Hickman & Hill 1978, Hocking *et al.* 1978, Jarman & Crowden 1978, Rose 1978; the Warra Silvicultural Systems Trial (Neyland *et al.* 2012); and the Wilderness Ecosystems Baseline Studies (Driessen *et al.* 1998)) have generated a wealth of information and specimens that have led to the documentation of new species. However, apart from past *Bush Blitz* surveys, no previous work has aimed to document both the flora and fauna so comprehensively at a particular location or, most importantly, has been backed up with a more-or-less complete collection of the voucher material that is so critical for the identification process and which underpins future research (Hüber 1998, Culley 2013). With estimates suggesting that approximately 70% of Australian organisms are yet to be discovered and described (Cassis *et al.* 2016), it is only to be expected that with concerted searches new species will continue to be found. Whilst this is not true for all groups of organisms, such as the well-documented and highly visible mammals, birds and flowering plants, it is still very much the case for lesser-known or less conspicuous groups such as lichens and invertebrates. Certainly, surveys of these organisms undertaken to date indicate that Tasmania is rich in novelties.

The *Wind Song* property on Tasmania's East Coast was chosen for the first of these expeditions as it is known to contain a diverse range of habitats, it was readily accessible from Hobart, and the project had the enthusiastic support of the landowners. Drawing on the specialist expertise of TMAG staff and associates, the survey focused on vascular plants, bryophytes, lichens, butterflies, moths, beetles, snails and slugs; other taxonomic groups were also recorded opportunistically. Here we present an inventory and discussion of the plants, lichens and animals discovered. As well as offering an insight into a biodiverse corner of Tasmania, the results serve as a benchmark for future studies in other parts of Tasmania.

MATERIALS AND METHODS

The property

Wind Song is a 220 ha former farming property, situated at Little Swanport on Tasmania's east coast (pl. 1). The property is bounded by Swanston Road along the northern margin, by Strip Road along the western margin, and by private property along most of its other boundaries. It is intersected by the intermittently flowing White Hut Creek. The property has a shallow elevation profile, with the flats along the course of White Hut Creek situated at approximately 10 m a.s.l., and the northwest corner at approximately 70 m a.s.l. Much of the wooded area is protected by a 42 ha private reserve (pl. 1).

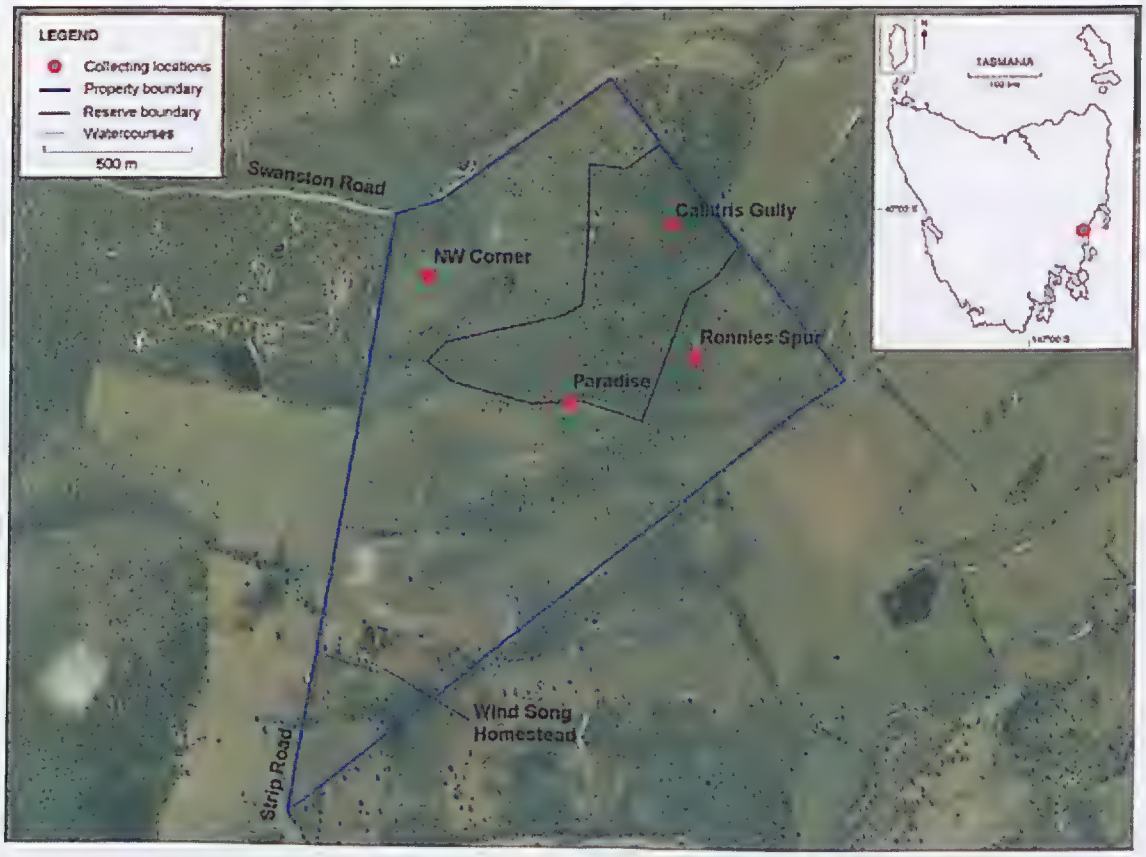


PLATE 1 – Location of *Wind Song*, Tasmania: showing main collecting locations and private reserve boundary.

Geologically, the property straddles the boundary between Triassic sandstone (with siltstone and mudstone), which underlies most of the property's surface, and Jurassic dolerite, which outcrops along the western margin (Calver *et al.* 2016). The sedimentary sequences are partly metamorphosed near the geological boundary, with patches of hornfels exposed near the northern edge of the property. Some of the northwesterly to southeasterly orientated spurs contain abundant pisolitic ironstone and represent relict Tertiary laterites.

The climate in central eastern Tasmania where the property is situated has a Köppen classification of *Cfb* (temperate, warm summer, without dry season (Peel *et al.* 2007)). Average temperatures in the nearby town of Orford fluctuate from a winter minimum of 3.5°C and maximum of 13°C in July, to a summer minimum of 12°C and maximum of 22°C in February. Rain is more common in late winter and spring, with an annual average of 668 mm (BOM 2018).

There is a history of sheep grazing and timber harvesting on the property, but these activities have largely ceased and stocking levels were greatly reduced approximately seven years ago. A small number of sheep and horses remain on a portion of the property and are used primarily for land management and recreational purposes. Occasional firewood collection continues.

Mapped vegetation types on *Wind Song* include the following: *Eucalyptus globulus* dry forest and woodland (DGL); *Eucalyptus pulchella* forest and woodland (DPO); *Pteridium esculentum* fernland (FPF); regenerating cleared land (FRG); and agricultural land (FAG) (Kitchener & Harris 2005).

Prior to this survey, limited data on the flora and fauna of the property were available in the form of an unpublished list (Lloyd unpublished), and via some records on the *Atlas of Living Australia* (Anonymous 2018). These data sources listed 120 vascular plant species, three invertebrates, three frogs, 34 birds and one reptile. Notable species previously recorded are the Critically Endangered Swift Parrot (*Lathamus discolor*) and the Endangered Chaostola Skipper (*Antipodia chaostola leucophaea*).

Expertise and timing

The project involved four botanists and three zoologists from TMAG staff, as well as two honorary zoologist researchers. The property was surveyed during 23–27 October 2017, with follow-up invertebrate sampling and trap-sample collection on 14 November and 12 December 2017, and further flora sampling on 7 March 2018 and 26 September 2018.

Site selection

Survey sites were selected in order to represent the major habitat types on the property. The sites were confined mostly to the de-stocked and regenerating portions. Areas with large infestations of Gorse (*Ulex europaeus*) and areas of primarily exotic pasture were largely excluded from the

survey. A summary of the main collecting areas is given in table 1 and plate 1. Surveys for lichens and bryophytes were based chiefly on detailed examination of three sites (Ronnies Spur; Callitris Gully and its margins; and NW Corner), supplemented by a cursory scan of some other parts of the property. Invertebrate specimens were collected from many locations across the property, although general insect trapping focused on Callitris Gully and the vicinity of Paradise.

TABLE 1 – Major collecting sites at *Wind Song*.

Site	Latitude	Longitude
Ronnies Spur	42°21'16.34"S	147°55'07.81"E
Callitris Gully	42°21'00.81"S	147°55'06.28"E
NW Corner	42°21'05.62"S	147°54'32.20"E
Paradise	42°21'22.2"S	147°54'39.8"E

Sampling methods

Specimens of vascular plants, bryophytes and lichens were collected and lodged in the Tasmanian Herbarium (HO), with limited duplicate specimens distributed to other herbaria nationally and internationally under TMAG's formal specimen exchange program. Several vascular plant taxa were recorded only by observation due to sampling difficulties (e.g., tall eucalypt trees) or lack of fertile material. All possible substrata for lichens and bryophytes, including rocks, soil, bark, wood and charcoal, were examined.

Moths were collected mainly by using ultraviolet light-traps. Both white sheets and bucket traps were used, but some were also collected from malaise traps (pl. 2A) set for general insect-sampling. Beetles, other insects and other arthropods were sampled through a mix of direct observation, hand collection, including the use of hand-nets and a beating-tray, and trapping, and as a by-product of light-trapping for moths; the traps employed were malaise traps, pitfall traps, trunk window traps and yellow pan traps (pl. 2 A–D). Molluscs were recorded through hand-searching and collection. Other invertebrates and vertebrates were recorded incidentally while searching for and collecting target taxa. Specimens were lodged in the TMAG Zoology collections, with some mollusc specimens temporarily retained in the private collection of Kevin Bonham.

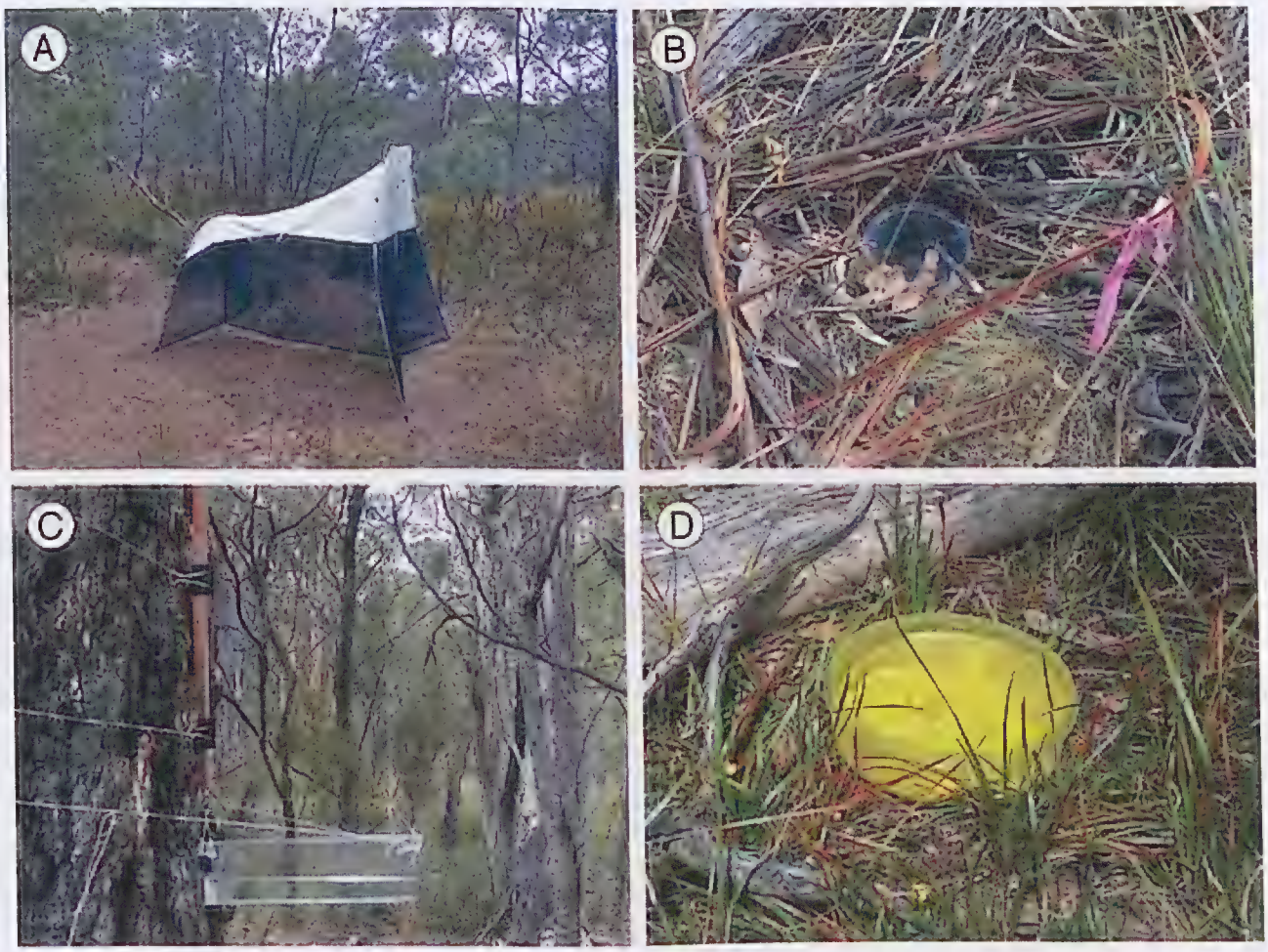


PLATE 2 – Insect traps used at *Wind Song*. (**A**) malaise trap. (**B**) pitfall trap. (**C**) trunk window trap. (**D**) yellow pan trap.

Specimen information from all survey material accessioned into the TMAG collection will be made available on the *Australasian Virtual Herbarium* and/or the *Atlas of Living Australia* in due course.

Specimen identification

Most specimens were identified with the aid of standard laboratory equipment and techniques. Where required, the reference collection of TMAG was used to check identifications. Lichens were identified in the laboratory by examination using low- and high-magnification of hand-cut sections of the thallus (vegetative tissue) and apothecia (reproductive structures), mounted in water, 10% KOH, 50% HNO_3, lactophenol cotton blue, ammoniacal erythrosin and Lugol's iodine. Routine chemical analyses using thin-layer chromatography followed standard methods (Orange *et al.* 2010). Some moth specimens were identified using the reference collections of the Australian National Insect Collection (ANIC) (CSIRO, Canberra) and the Biosecurity Tasmania Insect Collection at the New Town Research Laboratories of the Department of Primary Industries, Parks, Water and the Environment.

Nomenclature and distribution

Vascular plant nomenclature follows de Salas & Baker (2017). Nomenclature of vegetation types and their acronyms follows *TASVEG* (Harris & Kitchener 2005). Nomenclature for mosses and liverworts is in accordance with the *Australian Moss Name Index* (ABRS 2018a), the *Checklist of Australian Liverworts & Hornworts* (McCarthy 2006) and *Tropicos* (Tropicos.org. 2018). Lichen nomenclature mainly follows McCarthy (2018). Nomenclature for land snails follows Stanisic *et al.* (2018). For all other vertebrate and invertebrate taxa identified to species, nomenclature follows the *Australian Faunal Directory* (ABRS 2018b).

Undescribed or new species of moths are annotated with a unique phrase-name such as, '*Phaos* sp. 'BBTarkFish12". If specimens can be associated with previously collected material, already-existing epithets are adopted. Insect specimens that could only be identified to a taxonomic level higher than species are annotated with 'unplaced'.

Moth distributions and rarity were determined, in part, by referring to specimens in ANIC.

RESULTS

Diversity

This survey resulted in 886 taxa being recorded from *Wind Song*. Sixty-three taxa (three butterflies, 48 vascular plants, 11 birds and one reptile) had been recorded previously from the property but were not observed during this expedition (Anonymous 2018, Lloyd unpublished). If these unvouchered records are included then the total number of taxa for *Wind Song* is 949 (table 2, appendix 1). Several new species and new records for Tasmania, chiefly of lichens and invertebrates, were recorded and are detailed below.

One-hundred and three taxa were observed during this survey but were not collected. These taxa consisted of vertebrate fauna (40 birds, two frogs, five mammals and three reptiles), non-target invertebrates (39 taxa) and vascular plants (14 taxa).

Thirty-seven introduced species were recorded from the property. Twenty of these were vascular plants, five were birds and the remainder were invertebrate taxa referred to various groups.

Vegetation

The property comprises two broad habitat types – regenerating former pasture and woodland. Regenerating former pasture, once used for sheep grazing and largely devoid of trees, was typically associated with a ground cover of heavily grazed grasses and herbs, and dominated by sedges (including *Gahnia* spp., *Lepidosperma* spp., and *Lomandra longifolia*), copses of Silver Wattle (*Acacia dealbata* subsp. *dealbata*) and infestations of Gorse.

The following five vegetation communities occurred on the former pasture:

- dense sedgeland of *Lomandra longifolia* and *Lepidosperma* species with minor pasture grasses;
- Restionaceae- and Cyperaceae-dominated sedgeland in poorly drained areas containing species of *Lepidosperma, Gahnia, Juncus, Baumea, Tetraria* and *Schoenus* (pl. 3);
- pasture grassland comprising mostly exotic grasses (*Aira caryophyllea, Anthoxanthum odoratum* and *Vulpia* species) with occasional native species (*Themeda triandra, Poa labillardierei* and *Rytidosperma* species);
- *Leptospermum scoparium*-dominated sparse heathland;
- *Ulex europaeus*-dominated shrubland, in places forming an impenetrable thicket of almost 100% cover (pl. 4).

The remainder of the property consisted of *Eucalyptus*-dominated woodland with an understorey of low shrubs, tussock-forming monocotyledons and Silver Wattle, as well as small, discrete populations of Dogwood (*Pomaderris* spp.) and Oyster Bay Pine (*Callitris rhomboidea*).

Within the woodland, the following four main communities were recognised:

- *Eucalyptus globulus* and *E. viminalis* woodland, with an understorey of *Acacia dealbata* and *A. mearnsii*, occasional *Banksia, Allocasuarina* and *Leptospermum*, and a ground cover of *Lomandra longifolia* (pl. 5);
- *Acacia dealbata*- and *A. mearnsii*-dominated regenerating woodland with a sparse understorey and grass ground cover (one location supporting this habitat was called Paradise);
- *Eucalyptus globulus* gully woodland with a dense understorey of *Callitris rhomboidea* and *Pomaderris apetala*, occasional *P. elliptica* var. *diemenica, Bursaria spinosa* and *Dodonaea viscosa* subsp. *spathulata*, and a ground cover of *Lepidosperma* and *Lomandra* (the location supporting this habitat was named Callitris Gully) (pl. 6);

TABLE 2 – Overview of taxa recorded and collected from *Wind Song*.

Group	Total species	Total specimens	Introduced species
Vascular Plants	(190)	137	20
Dicotyledons	120	–	14
Gymnosperms	1	–	–
Monocotyledons	59	–	6
Pteridophytes	10	–	–
Bryophytes	(53)	58	–
Liverworts	13	–	–
Mosses	40	–	–
Lichens	170	212	
Invertebrates – Lepidoptera	165	755	1
Invertebrates – Coleoptera	105	280	1
Invertebrates – Other Insects	(165)	414	2
Blattodea	4	–	–
Dermaptera	1	–	–
Diptera	60	–	–
Hemiptera	32	–	–
Hymenoptera	–	–	2
Mantodea	–	–	–
Mecoptera	–	–	–
Neuroptera	–	–	–
Odonata	2	–	–
Orthoptera	13	–	–
Phasmida	1	–	–
Invertebrates – Other Arthropods	(19)	–	4
Acari	1	–	1
Araneae	13	–	–
Chilopoda	1	–	–
Diplopoda	1	–	1
Isopoda	2	–	2
Scorpiones	1	–	–
Invertebrates – Gastropoda	15	30	4
Invertebrates – Other	(3)	–	–
Annelida	1	–	–
Platyhelminthes	2	–	–
Vertebrates	(63)	–	5
Birds	51	–	5
Frogs	3	–	–
Mammals	5	–	–
Reptiles	4	–	–
Total	**949**	**1886**	**37**

- *Eucalyptus globulus* and *E. viminalis* tall woodland, with a highly reduced understorey of *Allocasuarina littoralis* and *Exocarpos cupressiformis* and an extremely sparse vascular plant ground cover (the location supporting this habitat was called Ronnies Spur). Occasional plant fossils were found at this location (pl. 7).

Evidence of heavy grazing pressure from native marsupials was widespread, with many Bennetts Wallabies (*Macropus rufogriseus*) and abundant marsupial scats observed. Former pasture was dominated by tussocks of robust, fibrous and unpalatable sedges, whereas palatable herbaceous species (e.g., *Lobelia anceps* and *Geranium* spp.) were largely restricted to the shelter inside tussocks or under the branches of fallen trees, where they were physically beyond the reach of grazing animals.

Gorse was by far the most common and widespread naturalised plant on the property and formed very large and dense infestations at several locations. Other widespread and common introduced plants included Sweet Vernal Grass (*Anthoxanthum odoratum*) and the herbaceous daisies, Cat's Ear (*Hypochaeris radicata*) and Hawkbit (*Leontodon saxatilis*).

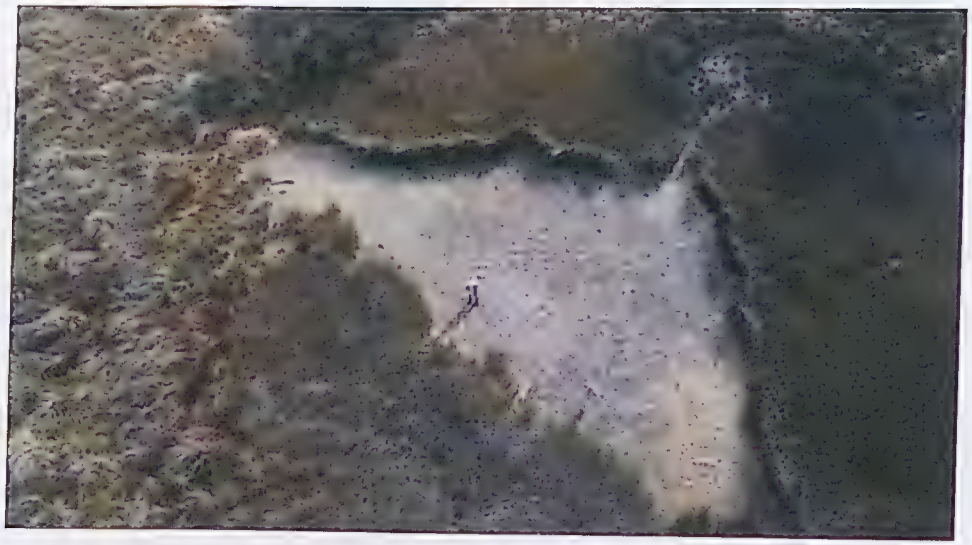


PLATE 3 – Aerial view of a typical Restionaceae- and Cyperaceae-dominated sedgeland at *Wind Song*.

PLATE 4 – Formerly grazed pasture forming a mosaic of introduced grasses, *Ulex europaeus* (dark green shrubs), *Lomandra longifolia*, *Gahnia* and native grass tussocks and copses of *Acacia dealbata* subsp. *dealbata* and surrounded by neighbouring sheep grazing properties in the middleground.

PLATE 5 – *Eucalyptus globulus* and *E. viminalis* woodland with an understorey of *Acacia dealbata* and *A. mearnsii*, containing occasional *Banksia*, *Allocasuarina* and *Leptospermum* and an abundant ground cover of *Lomandra longifolia*.

PLATE 6 – View of Callitris Gully looking east with *Callitris rhomboidea* (conical trees) dominant on the south facing slope.

PLATE 7 – Aerial view of Ronnies Spur: *Eucalyptus globulus* and *E. viminalis* tall woodland featuring very little ground cover.

Lichens were well represented in the woodland communities, and especially in the *Callitris*-dominated gully. They were less abundant in sedgy and shrubby former pasture vegetation, with the exception of occasional clumps of species of *Cladonia*. However, large rock outcrops throughout the property, both in woodland and pasture, supported rich assemblages of lichens, chiefly crustose species or foliose members of the Parmeliaceae. In woodland communities, lichens colonised most available substrates except for the eucalypts. These tend to support very few species, except on their basal stockings of stable bark, or on old fire scars of bleached wood or charcoal. Open ground in woodland was colonised by patches of *Heterodea muelleri* (pl. 8A).

Two general habitats for bryophytes were identified: open, dry and exposed sites, where they were growing predominantly on soil and rock (e.g., Ronnies Spur); and damper habitats close to watercourses, where they grew mainly on soil, humus and rock (e.g., Callitris Gully). Conditions at *Wind Song* were generally too dry for epiphytic bryophytes. However, one species of moss, *Rhaphidorrhynchium amoenum* var. *amoenum*, was found on bark on the lower trunk of *Pomaderris* in a damp area next to the creek at Callitris Gully. Another moss that usually grows as an epiphyte, *Zygodon intermedius*, was found on rock at the same location. The liverwort *Telaranea tasmanica* was the only Tasmanian endemic bryophyte recorded.

Fauna

Invertebrates – Lepidoptera (moths and butterflies)

The Lepidoptera fauna was generally typical of species found during spring in dry sclerophyll woodland. For example, *Phelotis cognata* was commonly collected. This species feeds on Native Cherry (*Exocarpos cupressiformis*), a common tree in dry sclerophyll forest and widespread at *Wind Song*. No Tasmanian endemic species were recorded, probably because the vegetation types of the property were largely typical of the more general southern Australian flora.

Two sites with a high species diversity, with around 70 taxa each, were in eucalypt woodland (community type 1, see *Vegetation*, above) at NW Corner and Paradise. Many species at these locations feed on *Eucalyptus* and *Acacia*, host plants which have strong affiliations with Australian Lepidoptera.

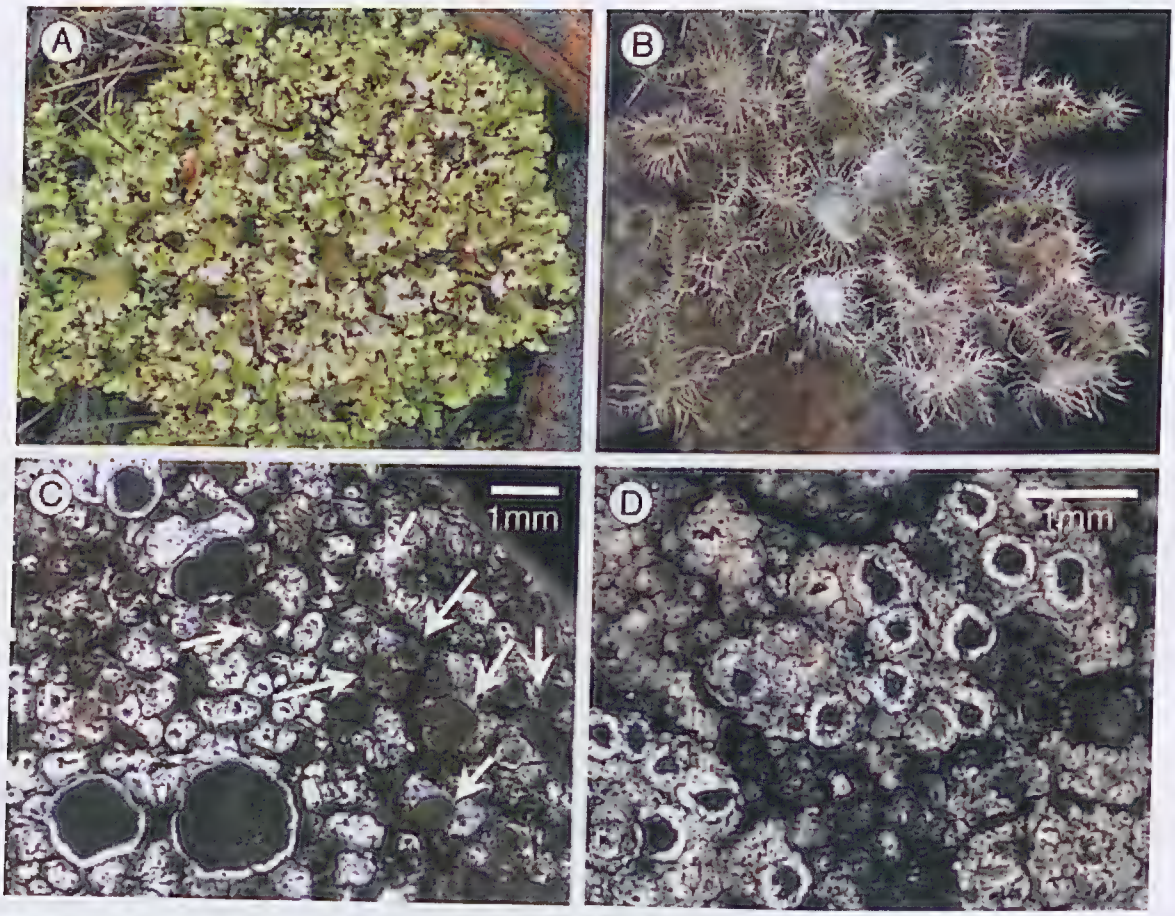


PLATE 8 – (**A**) *Heterodea muelleri*, a characteristic ground-dwelling lichen in dry sclerophyll woodlands with a very open understorey and ground layer. When dry, this species is difficult to spot as it appears as inconspicuous, shrivelled, brownish clumps. When moistened, its lobes unfurl to form attractive, bright green, lettuce-like growths on the forest floor. (**B**) *Usnea scabrida* subsp. *scabrida*, one of several species recorded for the first time for Tasmania. It grows on the twigs of Oyster Bay Pine, and is known from a few scattered locations along the East Coast. (**C**) *Caloplaca* sp. Seen as dull orange fruiting bodies (arrows), this remarkable find represents a species new to science. It has a unique ecology, growing as a parasite on the lichen *Tephromela atra*, seen here as the whitish thallus with large blackish apothecia with a white rim. (**D**) *Rinodina teniswoodiorum*, a new species discovered and described as a result of the *Wind Song* survey.

Highlights of the survey included the collection of one new species (see below under 'Novelties') and several species that are uncommon in Tasmania. The southeastern Australian micromoth *Gyphipterix cometophora*, previously known from just one specimen in Tasmania, was collected several times. Three uncommon southeastern Australian tortricid species were also collected, including *Thrincophora lignigerana* (pl. 9A). The occurrence of this species in Tasmania is otherwise only documented from Freycinet Peninsula in 1963 (ANIC). The others, *Euphona euphona* and *Anisogona mediana*, appear to be quite widespread in Tasmania, but have not been recorded in the state since the 1960s (ANIC), except for two records of *E. euphona* from Bruny Island in 2016 (Byrne 2017). Collection of the undescribed crambid species *Glaucocharis* ANIC sp. 10 is noteworthy. This species is only known from collections dating from the late 1970s from Westbury (ANIC). The southeastern Australian undescribed *Philobota* sp. ANIC66 (pl. 9B) was widespread across the property. This taxon is considered widespread in Tasmania but has likewise not been collected since the 1970s (ANIC). *Lecithocera terrigena* is one of the few members of the Lethoceridae family that occur in southern Australia. Previously it was only known from two collecting events in Tasmania (Byrne 2017), but it was collected twice from *Wind Song*. One specimen of the poorly known and uncommon *Antasia flavicapitata* was collected.

Many large flying insects appear in Tasmania as a result of being transported by strong northerly winds from mainland Australia. Two species of strong-flying moths in this category were collected during the survey. *Crioa hades* is a species of catocaline moth (underwing moths) that is not known to breed in Tasmania. The second species, *Aedia leucomelas* (eastern alchemist), is a globally ubiquitous noctuid moth. Both species are thought to be vagrants in Tasmania.

Invertebrates – Coleoptera (beetles)

Most beetle species found during this survey can be regarded as typical of dry woodland, or of associated localised habitats such as creeksides and seasonal waterbodies. Two notable leaf-beetles present on the property are the *Juncus*-associated

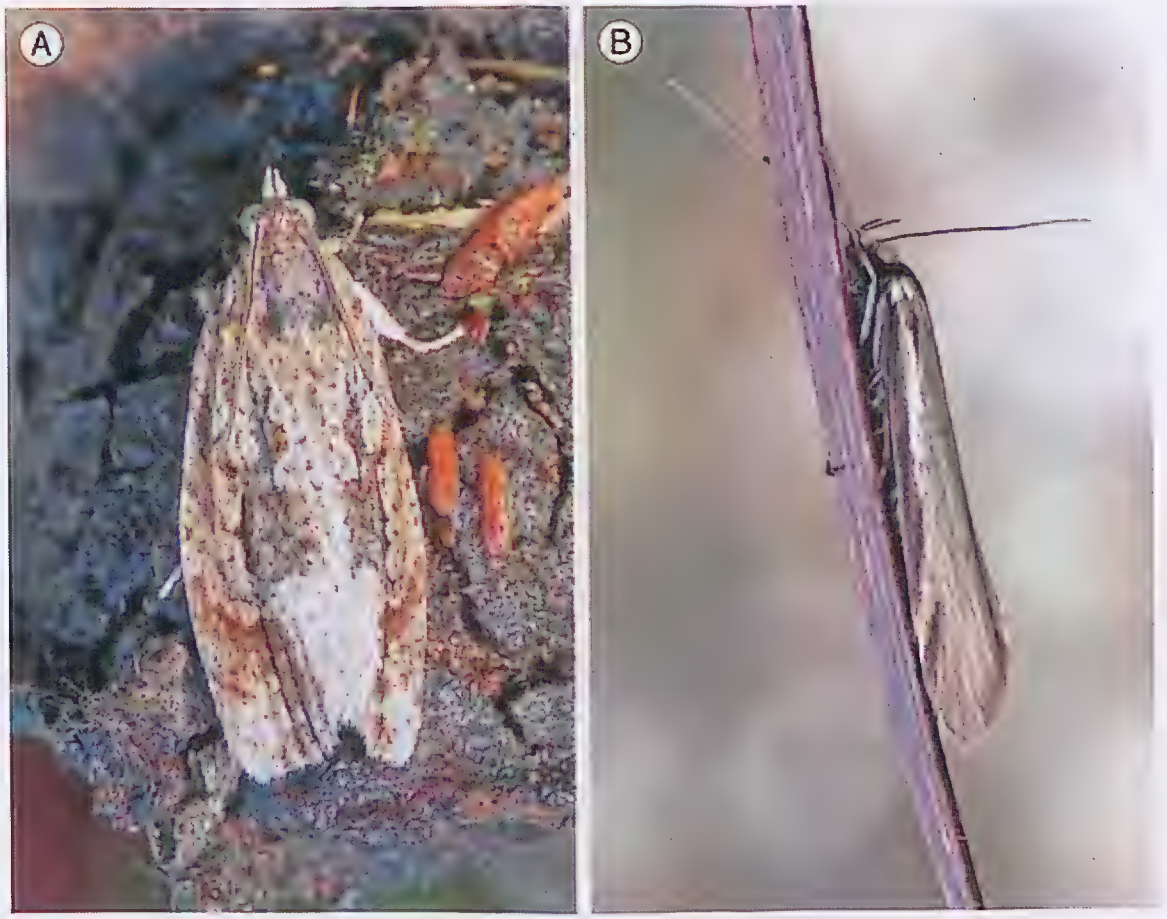


PLATE 9 – Rare species of *Lepidoptera* collected from *Wind Song*. (**A**) *Thrincophora lignigerana* (Tortricidae). (**B**) *Philobota* sp. ANIC 66, an undescribed species of Oecophoridae.

Eurispa albipennis (pl. 10A) and the *Acacia*-associated *Peltoschema oceanica*. Several individuals of the former species were found in open pasture near a small dam close to the property's southeastern boundary on the inflorescences of their foodplant, *Juncus*. A single specimen of the latter species was swept from its foodplant, *Acacia*, in the same general area. Both species appear to be patchily distributed along the eastern seaboard of Australia but there are rather few Tasmanian records of either. One of the more common leaf-beetles on eucalypt foliage was the strikingly coloured *Paropsisterna nobilitata* (pl. 10B). The saproxylic beetle fauna, normally a significant component of woodland beetle diversity, was rather limited, presumably because of lack of available habitat. The most frequently observed species was the darkling-beetle (*Isopteron obscurum*) (pl. 10C).

Invertebrates – Other insects

Many additional invertebrates outside of the target groups were also recorded and/or collected. These include cockroaches, termites, earwigs, true flies, true bugs, ants and bees, hanging-flies, lacewings and antlions, mantises, damselflies and dragonflies, grasshoppers and crickets, and stick-insects. The saproxylic stiletto-fly species, *Agapophytus quatiens* (pl. 11A), whose larvae are thought to be predators of other insect larvae living within logs, was recorded within the reserved section of the property. The males of this species gather in 'leks' on fallen logs, drumming their front feet as part of their display aimed at defending mating territories. Six species of beefly were recorded in areas of woodland with extensive open ground beneath (such as Ronnies Spur). Beefly larvae are parasitoids of other insects, particularly solitary bees and wasps. One such species, the strikingly marked *Anthrax maculatus* (pl. 11B), is saproxylic, being associated with exposed, often burnt, dead wood. The White-lined Cricket (*Trigonidium albovittata*), a species that favours lowland dry woodlands, was one of three orthopteran species found among leaf-litter (pl. 11C). The cryptically coloured groundhopper species *Paratettix argillaceus* was found among short-grazed damp grassy depressions where it is thought to graze on algae and lichens (pl. 11D).

Invertebrates – Other Arthropods

Eighteen non-insect arthropods belonging to a range of taxonomic groups were recorded from the property. These included mites, spiders, scorpions, millipedes, centipedes and slaters. The tiny ant-eating spider, *Euryopis splendens*, was found in crevices of dry logs (pl. 12A).

Invertebrates – Gastropoda (snails and slugs)

The rather depauperate gastropod fauna, comprising eleven native and four exotic species, is consistent with the (formerly) disturbed and fragmented nature of much of the property.

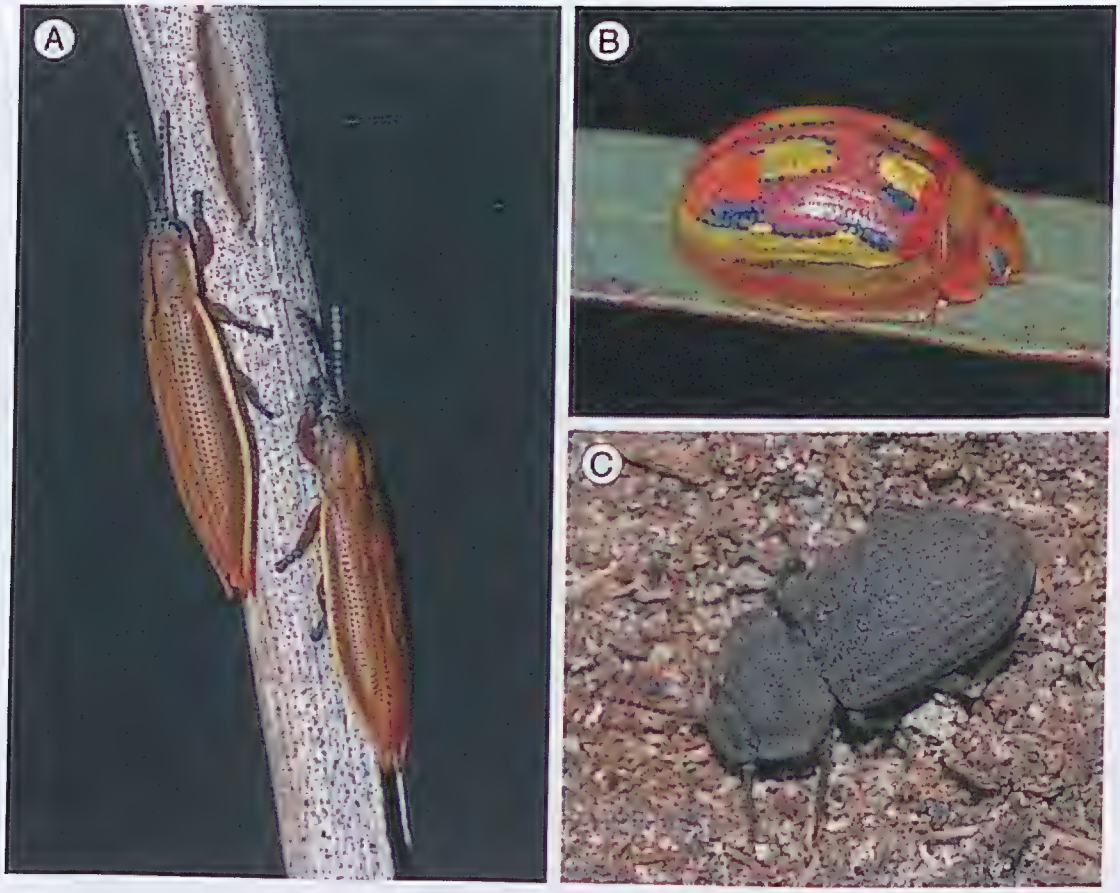


PLATE 10 – (**A**) The *Juncus*-associated leaf-beetle *Eurispa albipennis*. Length: 11 mm. (**B**) The eucalypt-associated leaf-beetle *Paropsisterna nobilitata*. (**C**) The saproxylic darkling-beetle *Isopteron obscurum*.

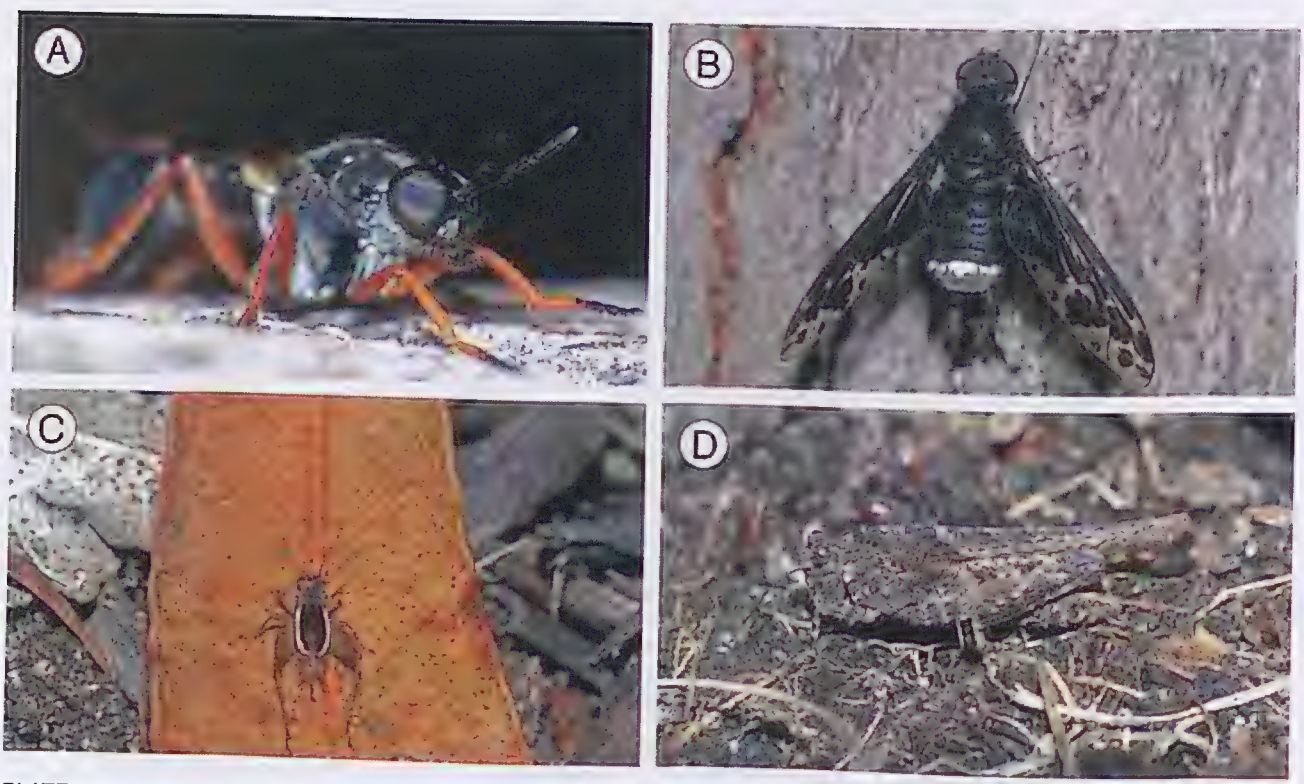


PLATE 11 – (**A**) The stiletto-fly *Agapophytus quatiens*. (**B**) The beefly *Anthrax maculatus*. (**C**) White-lined cricket *Trigonidium albovittata*. (**D**) The groundhopper *Paratettix argillaceus*.

Invertebrates – Other

Two species of terrestrial flatworm were found under rotting logs: one was provisionally identified as *Anzoplana trilineata* (pl. 12B) and the other as *Artioposthia nichollsi*. Tiger Leeches (*Philaemon grandis*) were numerous in damp areas (pl. 12C).

Vertebrates

While vertebrates were not specifically targeted in this survey, many species were observed and recorded. These comprised three species of frogs, three species of lizards, five species of mammals and 40 species of birds. All three frog species had been recorded in previous surveys of the property. One species of snake and eleven species of birds had also been recorded in previous surveys but were not encountered during this survey.

Novelties

Flora

Botanical novelties were limited to the lichens, of which a significant number was recorded. These included several species new to science, two of which have been formally described: *Anisomeridium disjunctum* (McCarthy & Kantvilas 2018) and *Rinodina teniswoodiorum* (pl. 8D) (Elix *et al.* 2019a). Further undescribed species were found in the genera *Caloplaca* (a remarkable lichenicolous species; pl. 8C), *Lecanora* and *Pertusaria*, but these require additional research work and will be dealt with in the future.

Many lichens previously unrecorded for Tasmania (McCarthy 2018) were also collected in the course of the survey. Some proved, on further study, to have been collected previously, and resided amongst unidentified lichen specimens in the Tasmanian Herbarium, whereas others were genuine discoveries from the *Wind Song* survey. Nine such new records have been formally reported and discussed elsewhere: *Buellia inturgenscens*, *B. schaereri*, *B. subadjuncta*, *B. suttonensis*, *Cyphelium trachylioides*, *Lecanora epibryon* subsp. *epibryon*, *Lepraria jackii*, *Ochrolechia africana* and *Rhizocarpon viridiatrum* (Elix *et al.* 2019b). A further ten are listed below, and additional new records that require further work for confirmation are highly likely to be found in the genera *Candelariella*, *Hertelidea*, *Schaereria*, *Schismatomma* and *Trapelia*.

Aspicilia caesiocinerea. A cosmopolitan species, collected from dolerite rocks in a dry stream bed. Classified by some authors (e.g., McCarthy 2018) in the genus *Circinaria*.

Specimen examined: Tasmania: Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2017, G.Kantvilas 340/17 (HO).

Bacidia stenospora. A relatively widespread epiphytic species in Tasmania, although not formally cited previously in the literature. Also known from New Zealand and Kangaroo Island, South Australia.

PLATE 12 – (**A**) Ant-eating spider *Euryopis splendens*. (**B**) A terrestrial flatworm, provisionally identified as *Anzoplana trilineata*. (**C**) tiger leech *Philaemon grandis*.

Selected specimens examined: Tasmania: Chain of Lagoons river mouth, 41°40'S 148°18'E, 1973, G.C.Bratt 73/725 & J.A. Cashin (HO); southern slope of South Sister, 41°32'S 148°10'E, 640 m alt., 2004, G.Kantvilas 377/04B (HO); summit of Mt Murray, 42°28'S 147°59'E, 315 m alt., 2006, G.Kantvilas 190/06 (HO); Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2018, G.Kantvilas 101/18 (HO).

Caloplaca lateritia. Widely scattered and locally very common on exposed rocks in paddocks, dry sclerophyll forest and heathland, especially in low rainfall areas. Similarly widespread on the Australian mainland.

Specimens examined: Tasmania: c. 3 km E of Broadmarsh, 42°41'S 147°09'E, 60 m alt., 1993, G.Kantvilas 158/93 & J.Elix (HO); Gowan Brae, eastern side of Nive River, 42°02'S 146°25'E, 810 m alt., 2014, G.Kantvilas 124/14, 133/14 (HO); Wind Song property, northern rim of Callitris Gully, 42°21'S 147°55'E, 60 m alt., 2017, G.Kantvilas 310/17 (HO); Wind Song property, Ronnies Spur, 42°21'S 147°55'E, 30 m alt., 2017, G.Kantvilas 263/17, 268/17 (HO).

Hypocenomyce tinderryensis. This species grows on eucalypt logs in dry sclerophyll forest together with the related and far more common and widespread *H. australis*. At *Wind Song* it was also found on old trunks of *Callitris rhomboidea*. Previously it was known only from mainland Australia.

Specimens examined: Tasmania: Bisdee Tier, 42°26'S 147°17'E, 640 m alt., 2009, G.Kantvilas 227/09 (HO); Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2018, G.Kantvilas 97/18 (CANB, HO, NY, UPS); Wind Song property, Paradise, 42°21'S 147°55'E, 30 m alt., 2018, G.Kantvilas 119/18 (HO).

Lecanora casuarinophila. Hitherto known only from mainland Australia (Lumbsch & Elix 2004) and Kangaroo Island (Kantvilas 2019), this species grows abundantly at the Wind Song property on the trunks of mature *Callitris rhombiodea*. This species may have been overlooked previously in Tasmania since apothecia occur infrequently.

Specimen examined: Tasmania: Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2018, G.Kantvilas 102/18 (HO).

Lecanora mobergii. Previously known from Western Australia and Kangaroo Island. This species is widespread in Tasmania on rocks in dry sclerophyll forest.

Specimens examined: Cherry Tree Hill along O Road, 41°58'S 148°08'E, 180 m alt., 2012, G.Kantvilas 330/12 (HO); Hellfire Bluff, below summit of westernmost high point, 42°44'S 147°55'E, 150 m alt., 2014, G.Kantvilas 373/14 (HO); Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2017, G.Kantvilas 335/17 (HO); Wind Song property, Ronnies Spur, 42°21'S 147°55'E, 30 m alt., 2017, G.Kantvilas 252/17 (HO).

Ochrolechia gyrophorica. Hitherto recorded only for mainland Australia. This species forms extensive, white, sorediate thalli on rough bark on the lower trunks of mature eucalypts. It has been collected infrequently, probably because usually it lacks fruiting bodies.

Specimens examined: Tasmania: Kellys Road, c. 1 km E of Mt Hobbs, 42°30'S 147°36'E, 520 m, 2018, G.Kantvilas 46/18 (HO); Wind Song property, Ronnies Spur, 42°21'S 147°55'E, 30 m alt., 2018, G.Kantvilas 117/18 (HO).

Ramboldia arandensis. This species is widespread on the southern Australian mainland (Elix 2009) where it grows on eucalypt wood. At *Wind Song* it was very abundant on the wood of *Callitris rhomboidea*.

Specimens examined: Tasmania: Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2017, G.Kantvilas 360/17, 364/17 (HO); Kellys Road, c. 1 km E of Mt Hobbs, 42°30'S 147°36'E, 520 m alt., 2018, G.Kantvilas 50/18 (HO).

Rinodia confusa. This species was previously considered endemic to South Australia. It was collected from the bark of *Callitris rhomboidea*.

Specimen examined: Tasmania: Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2017, G.Kantvilas 385/17 (HO).

Usnea scabrida* subsp. *scabrida. (pl. 8B) The genus *Usnea* poses many taxonomic problems in Australia and Tasmania, and the revision by Stevens (2004) did little to clarify the situation. These Tasmanian collections accord with Stevens' interpretation of *Usnea scabrida* subsp. *scabrida*, a species that lacks asexual propagules and is characterised by containing usnic acid and scabrosins. Although not recorded for Tasmania (McCarthy 2018), it has been collected several times along the East Coast in dry sclerophyll woodland and coastal scrub. Several collections are associated with *Callitris rhomboidea*, as is the collection from *Wind Song*.

Specimens examined: Tasmania: Point Meredith, 42°05'S 148°13'E, 10 m alt., 2002, G.Kantvilas 317/02 (HO); Yellow Sandbanks, Moulting Lagoon, 42°05'S 148°11'E, 2 m alt., 2004, G.Kantvilas 403/04 (HO); summit of Mt Murray, 42°28'S 147°59'E, 315 m alt., 2006, G.Kantvilas 183/06 (HO); Buxton River at the old weir, 42°15'S 147°59'E, 25 m alt., 2008, G.Kantvilas 136/08 (HO); S of Orford, 42°35'S 147°53'E, 130 m alt., 2011, G.Kantvilas 238/11 (HO); Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2017, G.Kantvilas 390/17 (HO).

Fauna

Many insect specimens collected remain unidentified or only partially identified (e.g., to family or genus level). It is possible that some of these represent undescribed species, but to determine this would require a significant amount of research, some with external collaborators. In addition to the three putative new taxa collected during the survey, eight species of Lepidoptera known to be undescribed were recorded.

Pateena* sp. nr *polymitarior. The specimens of this tiny jumping soil-bug (pl. 13) represent a putatively undescribed species not previously collected (Lionel Hill pers. comm.). Several specimens of this taxon were extracted from yellow pan-traps in Callitris Gully. *Pateena* species are generally associated with higher altitudes or much wetter habitats, so their presence at *Wind Song* is surprising (Lionel Hill pers. comm.).

Specimens examined: Tasmania: Wind Song property, Callitris Gully, 42°21'S 147°55'E, 40 m alt., 2017, S.J. Grove, F47315, F47316, F47317 & F47318 (TMAG).

***Agriophara* BYRNE 'Wind Song sp. 01' (Oecophoridae: Stenomatinae).** This species of moth is likely to be new to science and was recorded from only one site. The larvae of many *Agriophara* species feed between joined leaves, mostly of *Eucalyptus*.

Specimens examined: Tasmania, Wind Song property, NW sector, 42°21'15.5"S 147°54'20.9"E, 2017, C.J. Byrne, F58472 (TMAG).

***Tasmathera* sp.** This apparently undescribed species of charopid snail is the most significant mollusc species recorded. It was recorded on the basis of a single long-dead and very damaged shell, so its identity could not be confirmed. The shell has an unusually wide and deep umbilicus and does not resemble other east-coast *Tasmathera* specimens; indeed, it seems closest to *T. legrandi* (Cox, 1868) from western-shore Hobart dry forests. Charopid snails are more environmentally sensitive than other native snails overall, so this specimen may be a relict from a population that has since become extinct. Further sampling of other more intact areas nearby will be required to confirm its status as a new species.

Specimens examined: Specimens of various *Tasmathera* species in K. Bonham's private collection; not yet registered into TMAG collections.

Threatened species

No formally listed threatened species of vascular plants, bryophytes or lichens were collected from *Wind Song*, although many of the non-vascular species are considered uncommon and could well qualify for listing.

The most notable threatened species observed during the survey was the Swift Parrot (*Lathamus discolor*), a species listed as Critically Endangered under the Commonwealth *Environment Protection and Biodiversity Conservation Act* (1999) and as Endangered under the *Tasmanian Threatened Species Protection Act* (1995). Individuals were seen or heard flying overhead on several occasions; they were also heard foraging in flowering Blue Gum (*Eucalyptus globulus*) trees on the property. It might nest on the property, although higher-quality nesting habitat probably exists elsewhere in the vicinity. The Wedge-tailed Eagle (*Aquila audax*) was also observed and may well nest on the property towards the eastern edge of the main woodland block where a nest was observed. This species is listed as Endangered under both Commonwealth and Tasmanian legislation.

The butterfly species of most note at *Wind Song* is the Chaostola Skipper (*Antipodia chaostola leucophaea*) which had been recorded previously through larval searches by Dr Phil Bell (pers. comm. Jane Teniswood). Since 2010, the species has been listed as Endangered under both Commonwealth and Tasmanian laws. Regrettably, no adults of this species were found, despite the first sampling period coinciding with the usual adult flight period; this is not atypical for populations of this species. However, the habitat appeared suitable, at least in places; the butterfly is associated with warm, open-grown but sheltered stands of *Gahnia* sedges chiefly *G. radula* (probably the larval food plant), generally on sandy soils. During the December visit, a small number of the Tasmanian Silky Hairstreak Butterfly (*Pseudalmenus chlorinda chlorinda*) (pl. 14) were found on older Silver Wattles (*Acacia dealbata*) within the main woodland block. This species is listed as Rare under the Tasmanian legislation.

PLATE 13 – The minute jumping-bug *Pateena* sp. nr *polymitarior*. Length: 1 mm.

PLATE 14 – Tasmanian silky hairstreak butterfly *Pseudalmenus chlorinda chlorinda*.

Exotic and pest species

The introduced vascular plants recorded for *Wind Song* consisted of species common to agricultural and urban areas of Tasmania. Two species, Gorse (*Ulex europaeus*) and Creeping Thistle (*Cirsium arvense* var. *arvense*) (pl. 15A), are declared weeds under the Tasmanian *Weed Management Act* (1999). These and the other species of introduced plants recorded are likely to have originated from the previous agricultural activities conducted on the property.

Several exotic insects, snails and other invertebrates were recorded: 11-spotted Ladybird (*Coccinella undecimpunctata*), European Honeybee (*Apis mellifera*), European Wasp (*Vespula germanica*), Cabbage White Butterfly (*Pieris rapae*), Cabbage Moth (*Plutella xylostella*), Gorse Spider-mite (*Tetranychus lintearius*) (pl. 15B), Portuguese Millipede (*Ommatoiulus moreleti*), European slaters (*Armadillidium vulgare* and *Porcellio scaber*), Hedgehog Slug (*Arion intermedius*), Garden Snail (*Cornu aspersum*), Striped Field-slug (*Lehmannia nyctelia*) and Garlic Snail (*Oxychilus alliarius*). Exotic birds recorded were: European Skylark (*Alauda arvensis*), European Goldfinch (*Carduelis carduelis*), Laughing Kookaburra (*Dacelo novaeguineae novaeguineae*, introduced to Tasmania from mainland Australia), Common Starling (*Sturnus vulgaris*) and European Blackbird (*Turdus merula merula*).

Ten native moth species considered to be agricultural pests were collected during the survey: Painted Apple Moth (*Orgyia anartoides*) (pl. 15C), *Pantydia sparsa*, Cherry Looper Moth (*Chloroclystis approximate*), Twig Looper (*Ectropis excursaria*), Brown Cutworm (*Agrotis munda*), Variable Cutworm (*Agrotis porphyricollis*), Eastern Alchemist (*Aedia leucomelas*), Lightbrown Apple Moth (*Epiphyas postvittana*), *Epiphyas xylodes* and *Acropolitis rudisana*. The presence of such a high number of lepidopteran pests is most likely a result of the modified agricultural environment prevalent in this area.

DISCUSSION

Typical species

The broader characteristics of the flora (including bryophytes and lichens) and fauna of *Wind Song* were found to be consistent with past land use, where clearing, grazing, wood-cutting and frequent fires have rendered much of the site relatively species-poor and supporting only widespread, generally ecologically tolerant taxa. This applied particularly to non-wooded areas previously cleared for pasture, where the vascular flora was dominated by exotic weedy species, and unpalatable tussocks of native monocots and where 'native' constituents, for example the lichens, were restricted to outcropping boulders, small stones, and the intervening consolidated soil, or to remnant trees, stumps and wood fragments.

In general, all groups of fauna found at *Wind Song* constituted species typical of dry woodland or of associated localised habitats such as creek verges and seasonal waterbodies, such as occur widely in eastern Tasmania and south-eastern Australia. The seasonal lepidopteran fauna was typical of those species found during spring in dry sclerophyll woodland. This collection included approximately 16% of Tasmania's known lepidopteran fauna, and equates to high species diversity for this area. However, any interpretations of diversity should be tempered with caution as comparative data on similar surveys are scarce. Also, surveys such as these can only account for a fraction of the fauna because moths and butterflies are seasonal, and only some spring-flying species were captured. Furthermore, weather has a large effect on the appearance of species; most lepidopteran adults will not fly during cold and/or windy conditions, as was the case for the first three days of the survey. The rather depauperate gastropod fauna is consistent with the disturbed and fragmented nature of much of the property.

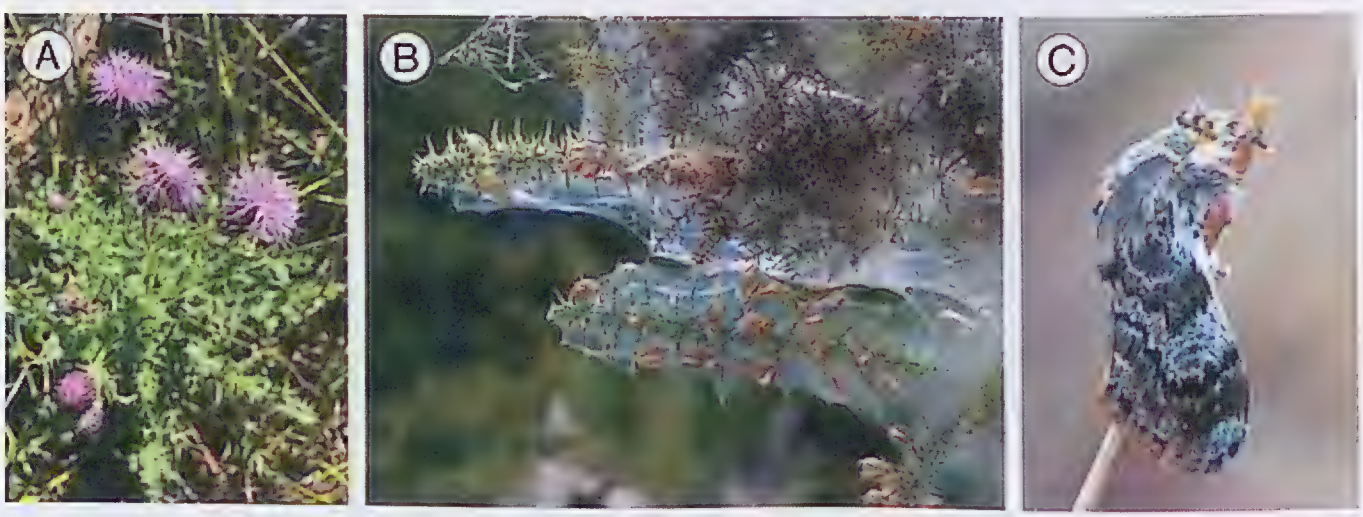


PLATE 15 – Introduced species recorded from *Wind Song*. (**A**) Creeping Thistle (*Cirsium arvense* subsp. *arvense*). (**B**) Webs of Gorse Spider-Mite (*Tetranychus lintearius*). (**C**) Painted Apple Moth (*Orgyia anartoides*) (Lepidoptera: Erebidae).

Species-rich areas

Two of the areas studied during the survey were of particular interest. The first, Callitris Gully, is a corridor of standing, mature Oyster Bay Pine (*Callitris rhomboidea*) with numerous fallen logs. While Oyster Bay Pine is a widespread tree on Tasmania's East Coast, 'stands' in which this species dominates are seen far less commonly. The stand at *Wind Song* is small and degraded, but was nevertheless of sufficient size to retain an interesting complement of lichen species on the oldest trunks and on some of the fallen logs. One of the new species described, *Anisomeridium disjunctum,* was found at this locality, as were several of the species newly recorded, notably the hitherto South Australian endemic, *Rinodina confusa*, as well as *Buellia schaereri*, *Ramboldia arandensis* and *Usnea scabrida* subsp. *scabrida*. Several of the lichens collected from this area are generally found in old-growth forest, not usually in dry sclerophyll communities. More than half of the moss species, and one third of the liverwort species, were found growing on rocks or soil in the damp conditions provided by the Gully. The tiny jumping soil-bug *Pateena* sp. nr *polymitarior* was also collected from this site. The *Callitris*-feeding geometrid moth *Corula geometroides* was also abundant at this site. This was also the main collecting locality for the following uncommon moths: *Glyphipteryx cometophora*, *Lecithocera terrigena* and *Anisogona mediana*. More than one-third of the Lepidoptera fauna was collected at this location.

The second locality of high biodiversity was the dry *Eucalyptus* woodland at Ronnies Spur. This site features outcrops of tertiary laterites that provided habitat for a surprisingly large number of ground-dwelling bryophytes and saxicolous lichens. The newly described lichen species, *Rinodina teniswoodiorum*, was collected here.

Management implications

Much of the *Wind Song* property has a history of extensive tree-clearing and grazing, and even the remnant areas retain a vegetation structure and species composition consistent with this past land-use. More recent fencing of the main blocks of woodland will have alleviated some of the grazing pressure from domestic stock; however, wallabies, occurring in high population densities within these fenced areas, have taken up much of the slack arising from de-stocking. Their abundance may be the chief reason for the relative lack of recent regeneration and recolonisation, particularly of palatable plant species. In turn, this limits the range of available feeding and nesting habitats and refugia for insects, birds (including Swift Parrot) and other animals. Historical firewood collection, involving the removal of fallen dead wood and the felling of dead or dying trees, also impacts on availability of present-day habitats. Dead wood, both on the ground and standing, is currently much sparser than would be the case in a less-impacted woodland, and this is reflected in the relatively depauperate associated fauna. The ecological recovery of these woodland blocks would be greatly aided by reducing the density of wallabies and by ceasing or limiting extraction of firewood. Further control of Gorse – at least limitation of its spread, and, if possible, its local eradication, is also highly recommended if considered feasible.

ACKNOWLEDGEMENTS

The Tasmanian Museum and Art Gallery's 2017 Expedition of Discovery would not have been possible without the generous support of Jane and Tom Teniswood and The Friends of the Tasmanian Museum and Art Gallery Incorporated. Keith Corbett is thanked for his remarks on the rock formation at Ronnies Spur. Gintaras Kantvilas acknowledges with many thanks the assistance of Jack Elix and Patrick McCarthy in identification of certain lichens, and Jean Jarman for preparation of the lichen plate.

REFERENCES

ABRS 2018a: *Australian Moss Name Index (AusMoss),* Australian Biological Resources Study, Canberra. Viewed 21 August 2018. https://moss.biodiversity.org.au/nsl/services/AusMoss.

ABRS 2018b: *Australian Faunal Directory.* Australian Biological Resources Study, Canberra. Viewed 15 December 2018. https://biodiversity.org.au/afd/mainchecklist.

Anonymous 2018: *Atlas of Living Australia.* Viewed 21 July 2018. http://www.ala.org.au.

Bauer, B., Mollison, R. & Young, C. 2010: *Bush Blitz, Tasmania 2010: Australian Biological Research Study Survey.* TMAG, Hobart.

BOM (Bureau of Meteorology) 2018: *Climate statistics for Australian locations: Orford (Aubin Court).* Viewed 20 August 2018. http://www.bom.gov.au/climate/averages/tables/cw_092027.shtml

Bratt, G.C. 1978: *Lower Gordon River Scientific Survey: Mosses and lichens.* The Hydro-Electric Commission, Hobart.

Byrne, C.J. 2017: *Bush Blitz Report – Bruny Island 15th–21st February 2016, Lepidoptera (excluding Papilionoidea). Report for the Bush Blitz Project,* Australian Biological Research Studies, CSIRO, ACT.

Calver, C.R., Corbett, K.D., Forsyth, S.M., Clark, M.J., McClenaghan, M.P. & Vicary, M. 2016: *Geology of Southeast Tasmania, Digital Geological Atlas 1:250,000 Scale Series, Version 6.* Viewed 21 August 2018. http://www.mrt.tas.gov.au/webdoc2/app/default/map_detail?id=898223

Cassis, G., Laffan, S.W. & Ebach, M.C. 2016: Biodiversity and Bioregionalisation Perspectives on the Historical Biogeography of Australia. *In* Ebach, M.C. (ed.): *Handbook of Australasian Biogeography.* Taylor & Francis, Melbourne.

Commonwealth of Australia 2012: *Egg Islands, Flat Rock, Lower Porter Hill and Porter Hill, Murphys Flat, Seventeen Mile Plain and Vale of Belvoir Reserves Tas, 2010, Bush Blitz Survey Report.* Australian Biological Resources Study, Canberra.

Commonwealth of Australia 2014a: *Skullbone Plains Tasmania 2012. A Bush Blitz Survey Report.* Australian Biological Resources Study, Canberra.

Commonwealth of Australia 2014b: *Flinders Island, Tasmania: Bush Blitz Species Discovery Program.* Australian Biological Resources Study, Canberra.

Commonwealth of Australia 2016: *Five Rivers Reserve and trawtha makuminya, Tasmania: Bush Blitz Species Discovery Program.* Australian Biological Resources Study, Canberra.

Commonwealth of Australia 2017a: *Southwest National Park Tasmania 2016. A Bush Blitz survey report.* Australian Biological Resources Study, Canberra.

Commonwealth of Australia 2017b: *Tarkine, Tasmania 2015. A Bush Blitz survey report.* Australian Biological Resources Study, Canberra.

Commonwealth of Australia 2017c: *Bruny Island Tasmania 2016. A Bush Blitz survey report.* Australian Biological Resources Study, Canberra.

Culley, T.M. 2013: Why vouchers matter in botanical research. *Applications in Plant Sciences*, **1**:11, apps.1300076. http://doi.org/10.3732/apps.1300076.

de Salas, M.F. & Baker, M.L. 2017: *A Census of the Vascular Plants of Tasmania & Index to the Student's Flora of Tasmania and Flora of Tasmania Online.* Tasmanian Herbarium, TMAG, Hobart. http://www.tmag.tas.gov.au

Driessen, M.M., Comfort, M.D., Jackson, J., Balmer, J., Richardson, A.M.M. & McQuillan, P.B. (eds) 1998: *Wilderness Ecosystems Baseline Studies (WEBS): Pelion Plains-Mt Ossa, 1990-1993. Tasmanian Parks and Wildlife Service Report 98/2.* Parks and Wildlife Service, Hobart.

Elix, J.A. 2009: Ramboldia. *Flora of Australia* **57**: 19–31.

Elix, J.A., McCarthy, P.M. & Kantvilas, G. 2019a: Two new species of *Rinodina* (Physciaceae, Ascomycota) from southern Australia. *Australasian Lichenology* **84**: 10–15.

Elix, J.A., McCarthy, P.M., Archer, A.W. & Kantvilas, G. 2019b: Additional lichen records from Australia. *Australasian Lichenology* **84**: 55–71.

Harris, S. & Kitchener, A. 2005: *From Forest to Fjaeldmark: Descriptions of Tasmania's Vegetation.* Department of Primary Industries, Water and Environment, Printing Authority of Tasmania, Hobart.

Hickman, J.L. & Hill, L. 1978: *Lower Gordon River Scientific Survey: Terrestrial invertebrates.* The Hydro-Electric Commission, Hobart.

Hocking, G.J., Guiler, E.R. & Colquhoun-Kerr, J.S. 1978: *Lower Gordon River Scientific Survey: The Mammals.* The Hydro-Electric Commission, Hobart.

Huber, J.T. 1998: The importance of voucher specimens, with practical guidelines for preserving specimens of the major invertebrate phyla for identification. *Journal of Natural History* **32**(3): 367–385, https:// doi.org/10.1080/00222939800770191.

Jarman, S.J. & Crowden, R.K. 1978: *Lower Gordon River Scientific Survey: a Survey of Vegetation.* The Hydro-Electric Commission, Hobart. 55 pp.

Kantvilas, G. 2019: An annotated catalogue of the lichens of Kangaroo Island, South Australia. *Swainsona* **32**: 1–97.

Kitchener, A. & Harris, S. 2013: *From Forest to Fjaeldmark: Descriptions of Tasmania's Vegetation. Edition 2.* Department of Primary Industries, Parks, Water and Environment, Tasmania.

Lumbsch, H.T. & Elix, J.A. 2004: Lecanora. *Flora of Australia* **56A**: 12–62.

McCarthy, P.M. 2006: *Checklist of Australian Liverworts and Hornworts.* Australian Biological Resources Study, Canberra. Viewed 21 August 2018. http://www.anbg.gov.au/abrs/liverwortlist/liverworts_a_z.html

McCarthy, P.M. 2018: *Checklist of the Lichens of Australia and its Island Territories.* Australian Biological Resources Study, Canberra. Version 17 May 2018. http://www.anbg.gov.au/abrs/lichenlist/introduction.html.

McCarthy, P.M. & Kantvilas, G. 2018: *Anisomeridium disjunctum* (Monoblastiaceae), a new lichen species from Tasmania, with a key to the genus in Australia. *Australasian Lichenology* **83**: 54–60.

Neyland, M., Hickey, J. & Read, S.M. 2012: A synthesis of outcomes from the Warra Silvicultural Systems Trial, Tasmania: safety, timber production, economics, biodiversity, silviculture and social acceptability. *Australian Forestry* **75**(3): 147–162.

Orange, A., James, P.W. & White, F.J. 2010: *Microchemical Methods for the Identification of Lichens.* British Lichen Society, London. 101 pp.

Peel, M.C., Finlayson, B.L. & McMahon, T.A. 2007: Updated world map of the Köppen-Geiger climate classification. *Hydrology and Earth Systems Sciences* **11**: 1633–1644

Rose, R. 1978: *Lower Gordon River Scientific Survey: The Birds.* The Hydro-Electric Commission, Hobart: 29 pp.

Stanisic, J., Shea, M., Potter, D. & Griffiths, O. 2018: *Australian Land Snails. Volume 2. A Field Guide to Southern, Central and Western Species.* Bioculture Press, Mauritius: 594 pp.

Stevens, G.N. 2004: Usneaceae. *Flora of Australia* **56A**: 78–115.

Taxonomy Decadal Plan Working Group 2018: *Discovering Biodiversity: A decadal plan for taxonomy and biosystematics in Australia and New Zealand 2018–2028.* Australian Academy of Science and Royal Society Te Apārangi, Canberra and Wellington

Tropicos.org. 2018: *Botanical Information System at the Missouri Botanical Garden.* (Missouri Botanical Garden, Saint Louis). Viewed 21 August 2018. http://www.tropicos.org

(Accepted 7 May 2019)

APPENDIX 1

Flora and fauna of *Wind Song* (Taxa marked with * were observed during the survey but not collected; i signifies a taxon that is introduced in Tasmania; e signifies a taxon that is endemic in Tasmania; $ signifies a taxon not recorded during this survey but recorded in past surveys).

Appendix 1.1: Vascular plant taxa of *Wind Song*

PTERIDOPHYTES

ADIANTACEAE

$ *Adiantum aethiopicum* L.

Cheilanthes austrotenuifolia H.M.Quirk & T.C.Chambers

ASPLENIACEAE

Asplenium flabellifolium Cav.

BLECHNACEAE

Blechnum nudum (Labill.) Mett. ex Luerss.

$ *Blechnum wattsii* Tindale

DENNSTAEDTIACEAE

Hypolepis glandulifera Brownsey & Chinnock

Pteridium esculentum (G.Forst.) Cockayne subsp. *esculentum*

GLEICHENIACEAE

Gleichenia microphylla R.Br.

LINDSAEACEAE

Lindsaea linearis Sw.

SELAGINELLACEAE

Selaginella uliginosa (Labill.) Spring

GYMNOSPERMS

CUPRESSACEAE

Callitris rhomboidea R.Br. ex Rich. & A.Rich.

MONOCOTYLEDONS

CENTROLEPIDACEAE

Centrolepis fascicularis Labill.

CYPERACEAE

Baumea acuta (Labill.) Palla

Baumea juncea (R.Br.) Palla

Baumea tetragona (Labill.) S.T.Blake

Carex breviculmis R.Br.

Gahnia radula (R.Br.) Benth.

Lepidosperma concavum R.Br.

Lepidosperma elatius Labill.

e *Lepidosperma globosum* Labill.

Lepidosperma gunnii Boeckeler

e *Lepidosperma inops* F.Muell. ex Rodway

Lepidosperma laterale R.Br.

Lepidosperma longitudinale Labill.

Schoenus apogon Roem. & Schult.

Tetraria capillaris (F.Muell.) J.M.Black

IRIDACEAE

Diplarrena moraea Labill.

Patersonia fragilis (Labill.) Asch. & Graebn.

JUNCACEAE

$ *Juncus gregiflorus* L.A.S.Johnson

Juncus kraussii Hochst. subsp. *australiensis* (Buchenau) Snogerup

Juncus pallidus R.Br.

Juncus subsecundus N.A.Wakef.

LILIACEAE

$ *Arthropodium milleflorum* (DC.) J.F.Macbr.

$ *Dianella revoluta* R.Br. var. *revoluta*

Hypoxis hygrometrica Labill. var. *hygrometrica*

$ *Pauridia vaginata* (Schltdl.) Snijman & Kocyan var. *vaginata* (Schltdl.) Snijman & Kocyan

Thysanotus patersonii R.Br.

$ *Wurmbea dioica* (R.Br.) F.Muell. subsp. *dioica*

ORCHIDACEAE

Cyrtostylis reniformis R.Br.

$ *Diuris pardina* Lindl.

* *Diuris sulphurea* R.Br.

Eriochilus cucullatus (Labill.) Rchb.f.

Glossodia major R.Br.

$ *Microtis parviflora* R.Br.

Pterostylis pedunculata R.Br.

Pterostylis curta R.Br.

POACEAE

Agrostis parviflora R.Br.

i *Aira caryophyllea* L. subsp. *caryophyllea*

i *Anthoxanthum odoratum* L.

Austrostipa pubinodis (Trin. & Rupr.) S.W.L.Jacobs & J.Everett

Echinopogon ovatus (G.Forst.) P.Beauv.

$i *Dactylis glomerata* L.

i *Holcus lanatus* L.

Lachnagrostis filiformis (G.Forst.) Trin.

Microlaena stipoides (Labill.) R.Br. var. *stipoides*

i *Phalaris aquatica* L.

$ *Poa labillardierei* Steud. var. *acris* Vickery

Poa labillardierei Steud. var. *labillardierei*

Poa rodwayi Vickery

Rytidosperma pilosum (R.Br.) Connor & Edgar

Rytidosperma racemosum (R.Br.) Connor & Edgar var. *racemosum*

* *Tetrarrhena distichophylla* (Labill.) R.Br.

Themeda triandra Forssk.

i *Vulpia bromoides* (L.) Gray

RESTIONACEAE

Apodasmia brownii (Hook.f.) B.G.Briggs & L.A.S.Johnson

$ *Baloskion australe* (R.Br.) B.G.Briggs & L.A.S.Johnson

Empodisma minus (Hook.f.) L.A.S.Johnson & D.F.Cutler

$ *Hypolaena fastigiata* R.Br.

Leptocarpus tenax (Labill.) R.Br.

XANTHORRHOEACEAE

Lomandra longifolia Labill.

DICOTYLEDONS

APIACEAE

Hydrocotyle hirta R.Br. ex A.Rich.

ASTERACEAE

Brachyscome spathulata Gaudich.

Cassinia aculeata (Labill.) R.Br. subsp. *aculeata*

* *Chrysocephalum apiculatum* (Labill.) Steetz subsp. *apiculatum*

i *Cirsium arvense* (L.) Scop. var. *arvense*

*i *Cirsium vulgare* (Savi) Ten.

$ *Coronidium scorpioides* (Labill.) Paul G.Wilson

$e *Craspedia glauca* (Labill.) Spreng.

Appendix 1.1 cont.

Euchiton japonicus (Thunb.) Holub
i *Hypochaeris radicata* L.
Lagenophora stipitata (Labill.) Druce
i *Leontodon saxatilis* Lam.
Leptorhynchos squamatus (Labill.) Less. subsp. *squamatus*
e *Olearia phlogopappa* (Labill.) DC. subsp. *gunniana* (DC.) Messina
$ *Olearia stellulata* (Labill.) DC.
$ *Senecio minimus* Poir.
$i *Sonchus asper* (L.) Hill
i *Taraxacum officinale* F.H.Wigg.
BORAGINACEAE
$ *Cynoglossum australe* R.Br.
CAMPANULACEAE
Lobelia anceps L.f.
Wahlenbergia gracilis (G.Forst.) A.DC.
CARYOPHYLLACEAE
Scleranthus biflorus (J.R.Forst. & G.Forst.) Hook.f.
i *Spergularia marina* (L.) Griseb.
CASUARINACEAE
Allocasuarina littoralis (Salisb.) L.A.S.Johnson
Allocasuarina verticillata (Lam.) L.A.S.Johnson
CHENOPODIACEAE
Einadia nutans (R.Br.) A.J.Scott subsp. *nutans*
CLUSIACEAE
Hypericum gramineum G.Forst.
CONVOLVULACEAE
Dichondra repens J.R.Forst. & G.Forst.
CRASSULACEAE
Crassula sieberiana (Schult. & Schult.f.) Druce
DILLENIACEAE
$ *Hibbertia hirsuta* (Hook.) Benth.
Hibbertia prostrata Hook.
Hibbertia riparia (R.Br. ex DC.) Hoogland
DROSERACEAE
Drosera auriculata Backh. ex Planch.
$ *Drosera peltata* Thunb.
Drosera pygmaea DC.
EPACRIDACEAE
$ *Acrotriche serrulata* (Labill.) R.Br.
* *Astroloma humifusum* (Cav.) R.Br.
$ *Brachyloma ciliatum* (R.Br.) Benth.
$ *Epacris impressa* Labill.
Epacris lanuginosa Labill.
$ *Leucopogon collinus* (Labill.) R.Br.
Leucopogon virgatus (Labill.) R.Br. var. *virgatus*
Lissanthe strigosa (Sm.) R.Br. subsp. *subulata* (R.Br.) J.M.Powell
$ *Styphelia adscendens* R.Br.
EUPHORBIACEAE
$ *Amperea xiphoclada* (Sieber ex Spreng.) Druce var. *xiphoclada*
$ *Beyeria viscosa* (Labill.) Miq.
Poranthera microphylla Brongn.
FABACEAE
$ *Bossiaea cinerea* R.Br.
Bossiaea prostrata R.Br.
$ *Daviesia ulicifolia* Andrews subsp. *ulicifolia*
$ *Glycine clandestina* J.C.Wendl.
$ *Gompholobium huegelii* Benth.
Hovea heterophylla A.Cunn. ex Hook.f.
$ *Indigofera australis* Willd. subsp. *australis*
$ *Kennedia prostrata* R.Br.
Pultenaea dentata Labill.
$ *Pultenaea juniperina* Labill.
Pultenaea pedunculata Hook.
Sphaerolobium minus Labill.
i *Ulex europaeus* L.
GENTIANACEAE
*i *Centaurium erythraea* Rafn
i *Cicendia filiformis* (L.) Delarbre
GERANIACEAE
Geranium potentilloides L'Hér. ex DC. var. *potentilloides*
$ *Geranium solanderi* Carolin
GOODENIACEAE
Goodenia lanata R.Br.
HALORAGACEAE
Gonocarpus micranthus Thunb.subsp. *micranthus*
Gonocarpus tetragynus Labill.
$ *Gonocarpus teucrioides* DC.
LINACEAE
Linum marginale A.Cunn.
MIMOSACEAE
Acacia dealbata Link subsp. *dealbata*
Acacia mearnsii De Wild.
Acacia melanoxylon R.Br.
e *Acacia mucronata* Willd. ex H.L.Wendl. subsp. *mucronata*
$ *Acacia myrtifolia* (Sm.) Willd.
$ *Acacia terminalis* (Salisb.) J.F.Macbr.
$ *Acacia verticillata* (L'Hér.) Willd. subsp. *verticillata*
MYRTACEAE
e *Eucalyptus amygdalina* Labill.
* *Eucalyptus globulus* Labill. subsp. *globulus*
* *Eucalyptus ovata* Labill. var. *ovata*
e* *Eucalyptus pulchella* Desf.
e *Eucalyptus viminalis* Labill. subsp. *viminalis*
Leptospermum lanigerum (Sol. ex Aiton) Sm.
Leptospermum scoparium J.R.Forst. & G.Forst.
$ *Melaleuca gibbosa* Labill.
e *Melaleuca virens* Craven
ONAGRACEAE
Epilobium billardiereanum Ser. ex DC. subsp. *billardiereanum*
OXALIDACEAE
* *Oxalis perennans* Haw
Oxalis rubens Haw
PITTOSPORACEAE
Bursaria spinosa Cav. subsp. *spinosa*
$ *Rhytidosporum procumbens* (Hook.) F.Muell.
PLANTAGINACEAE
i *Plantago coronopus* L. subsp. *coronopus*
Plantago varia R.Br.
POLYGALACEAE
Comesperma volubile Labill.
PRIMULACEAE
i *Lysimachia arvensis* (L.) U.Manns & Anderb.
PROTEACEAE
Banksia marginata Cav.
e* *Lomatia tinctoria* (Labill.) R.Br.
RANUNCULACEAE
$e *Clematis gentianoides* DC.
RHAMNACEAE
Pomaderris apetala Labill. subsp. *apetala*
e *Pomaderris elliptica* Labill. var. *diemenica* N.G.Walsh & Coates
$ *Pomaderris pilifera* N.A.Wakef. subsp. *pilifera*
ROSACEAE
Acaena novae-zelandiae Kirk
$ *Acaena ovina* A.Cunn.
Rubus parvifolius L.

Appendix 1.1 cont.

RUBIACEAE
* *Coprosma quadrifida* (Labill.) B.L.Rob.
Galium australe DC.
Galium gaudichaudii DC. subsp. *parviflorum* I.Thomps.
RUTACEAE
Zieria arborescens Sims subsp. *arborescens*
SANTALACEAE
Exocarpos cupressiformis Labill.
$ *Exocarpos strictus* R.Br.
Leptomeria drupacea (Labill.) Druce
SAPINDACEAE
Dodonaea viscosa Jacq. subsp. *spatulata* (Sm.) J.G.West
SCROPHULARIACEAE
Mazus pumilio R.Br.
*i *Verbascum thapsus* L.
$ *Veronica calycina* R.Br.
Veronica gracilis R.Br.
STYLIDIACEAE
* *Stylidium graminifolium* Sw.
THYMELAEACEAE
$ *Pimelea linifolia* Sm.
Pimelea humilis R.Br.
URTICACEAE
i *Urtica urens* L.
VIOLACEAE
Viola hederacea Labill. subsp. *hederacea*

Appendix 1.2: Bryophyte taxa of *Wind Song*

MOSSES
BARTRAMIACEAE
Breutelia affinis (Hook.) Mitt.
Breutelia sp.
BRYACEAE
Bryum clavatum (Schimp.) Müll.Hal.
Bryum microrhodon Müll.Hal.
Orthodontium lineare Schwägr.
Rosulabryum billardierei (Schwägr.) J.R.Spence
Rosulabryum torquescens (De Not.) J.R.Spence
DICRANACEAE
Campylopus insititius Hook.f. & Wilson
Campylopus introflexus (Hedw.) Brid.
Campylopus torquatus Mitt.
Dicranoloma billardierei (Brid.) Paris
DITRICHACEAE
Ceratodon purpureus (Hedw.)Brid.
Eccremidium pulchellum (Hook. & Wilson) Müll.Hal.
FISSIDENTACEAE
Fissidens oblongifolius Hook.f. & Wilson
Fissidens taylorii Müll.Hal.
Fissidens tenellus Hook.f. & Wilson
FUNARIACEAE
Entosthodon subnudus (Taylor) Fife var. *gracilis* (Hook.f. & Wilson) Fife
GRIMMIACEAE
Grimmia pulvinata (Hedw.) Sm. var. *africana* (Hedw.) Hook.f. & Wilson
Grimmia trichophylla Grev.
Schistidium apocarpum (Hedw.) Bruch & Schimp.
HEDWIGIACEAE
Hedwigia ciliata (Hedw.) P.Beauv.
Hedwigidium integrifolium (P.Beauv.) Dixon
HYPNACEAE
Hypnum cupressiforme Hedw.
LEMBOPHYLLACEAE
Lembophyllum clandestinum (Hook.f. & Wilson) Lindb. ex Paris
ORTHOTRICHACEAE
Zygodon intermedius Bruch & Schimp.
POLYTRICHACEAE
Polytrichum juniperinum Hedw.
POTTIACEAE
Barbula calycina Schwägr.
Syntrichia antarctica (Hampe) R.H.Zander
Triquetrella papillata (Hook.f. & Wilson) Broth.
Triquetrella tasmanica (Broth.) Granzow
Weissia sp.
PTYCHOMITRIACEAE
Ptychomitrium acutifolium Hook.f. & Wilson
Ptychomitrium mittenii A.Jaeger
PTYCHOMNIACEAE
Ptychomnion aciculare (Brid.) Mitt.
RACOPILACEAE
Racopilum cuspidigerum (Schwägr.) Ångstr. var. *convolutaceum* (Müll.Hal.) Zanten & Dijkstra
SEMATOPHYLLACEAE
Rhaphidorrhynchium amoenum (Hedw.) M.Fleisch. var. *amoenum*
Sematophyllum jolliffii (Hook.f.) Dixon
SPHAGNACEAE
Sphagnum novozelandicum Mitt.
THUIDIACEAE
Thuidiopsis furfurosa (Hook.f. &Wilson) M.Fleisch.
Thuidiopsis sparsa (Hook.f. & Wilson) Broth.

LIVERWORTS
ANEURACEAE
Riccardia sp.
AYTONIACEAE
Asterella drummondii (Hook.f. & Taylor) R.M.Schutst. ex D.G.Long
CEPHALOZIELLACEAE
Cephaloziella hirta (Steph.) R.M.Schust.
FRULLANIACEAE
Frullania falciloba Taylor ex Lehm.
Frullania probosciphora Taylor
GEOCALYCACEAE
Chiloscyphus novae-zelandiae (Lehm. & Lindenb.) J.J.Engel & R.M.Schust.
Chiloscyphus perpusillus (Hook.f. & Taylor) J.J.Engel
Chiloscyphus semiteres (Lehm. & Lindenb.) Lehm. & Lindenb.
Heteroscyphus ammophilus (Colenso) R.M.Schust.
Heteroscyphus knightii (Steph.) Grolle
LEPIDOZIACEAE
Kurzia sp.
Telaranea centipes (Taylor ex Gottsche, Lindenb. & Nees) R.M.Schust.
e *Telaranea tasmanica* (Steph.) J.J.Engel & G.L.Sm.Merrill

Appendix 1.3: Lichen taxa of *Wind Song*

Acarospora veronensis A.Massal.
Anisomeridium disjunctum P.M.McCarthy & Kantvilas
Arthonia sp. [HO 589535]
Aspicilia caesiocinerea (Nyl. ex Malbr.) Arnold
Austroparmelina conlabrosa (Hale) A.Crespo, Divakar & Elix
Austroparmelina labrosa (Zahlbr.) A.Crespo, Divakar & Elix
Austroparmelina pseudorelicina (Jatta) A.Crespo, Divakar & Elix
Bacidia bagliettoana (A.Massal. & De Not.) Jatta
Bacidia stenospora C. Knight
Bacidia sp. [HO 591827]
Baculifera xylophila (Malme) Marbach
Bapalmuia buchananii (Stirt.) Kalb & Lücking
Bryobilimbia australis (Kantvilas & Messuti) Fryday, Printzen & Ekman
Buellia austera Elix & Kantvilas
Buellia disciformis (Fr.) Mudd.
Buellia dissa (Stirt.) Zahlbr.
Buellia homophylia (C.Knight) Zahlbr.
Buellia inturgescens Müll.Arg.
Buellia mesospora Elix & Kantvilas
Buellia reagenella Elix
Buellia procellarum A.Massal.
Buellia schaereri De Not.
Buellia subadjuncta Elix & Kantvilas
Buellia suttonensis Elix & Knight
Buellia xanthonica (Elix) Elix
Byssoloma adspersum Malcolm & Vězda
Calicium glaucellum Ach.
Calicium victorianum (F.Wilson) Tibell subsp. *victorianum*
Caloplaca bartlettii S.Y.Kondr. & Kärnefelt
Caloplaca erythrosticta (Taylor) Zahlbr.
Caloplaca lateritia (Taylor) Zahlbr.
Caloplaca rexfilsonii S.Y.Kondr. & Kärnefelt
Caloplaca wilsonii S.Y.Kondr. & Kärnefelt
Caloplaca sp. [HO 590022]
? *Caloplaca* sp. [HO 590079]
Candelariella sp. [HO 589624]
Candelariella vitellina (Hoffm.) Müll.Arg.
Candelariella xanthostigmoides (Müll.Arg.) R.W.Rogers
Carbonea latypizodes (Müll.Arg.) Knoph & Rambold
Carbonicola foveata (Timdal) Bendiksby & Timdal
Chrysothrix xanthina (Vain.) Kalb
Cladia aggregata (Sw.) Nyl.
Cladia schizopora (Nyl.) Nyl.
Cladonia capitellata (Hook.f. & Taylor) var. *capitellata*
Cladonia capitellata var. *squamatica* A.W.Archer
Cladonia confusa R.Sant.
Cladonia corniculata Ahti & Kashiw.
Cladonia floerkeana (Fr.) Flörke
Cladonia humilis (With.) J.R.Laundon var. *humilis*
Cladonia merochlorophaea Asahina
Cladonia neozelandica Vain. var. *wilsonii* (A.W.Archer) Kantvilas
Cladonia paeminosa A.W.Archer
Cladonia pleurota (Flörke) Schaer.
Cladonia praetermissa A.W.Archer var. *praetermissa*
Cladonia pyxidata (L.) Hoffm.
Cladonia ramulosa (With.) J.R.Laundon
Cladonia rigida (Hook.f. & Taylor) Hampe var. *rigida*
Cladonia tenerrima (Ahti) S.Hammer
Cladonia verticillata (Hoffm.) Schaer.
Cyphelium trachylioides (Nyl.) Erichsen ex Keissel.
Diploschistes sticticus (Körb.) Müll.Arg.
Flavoparmelia haysomii (C.W.Dodge) Hale
Flavoparmelia rutidota (Hook.f. & Taylor) Hale
Halecania subsquamosa (Müll.Arg.) van den Boom & H.Mayrhofer
Hertelidea sp. [HO 589588]
Hertelidea pseudobotryosa R.C.Harris, Ladd & Printzen
Heterodea muelleri (Hampe) Nyl.
Hypocenomyce australis Timdal
Hypocenomyce scalaris (Ach.) M.Choisy
Hypocenomyce tinderryensis Elix
Hypogymnia billardierei (Mont.) Filson
Hypogymnia pulverata (Nyl.) Elix
Hypotrachyna revoluta (Flörke) Hale
Lecanora casuarinophila Lumbsch
Lecanora epibryon (Ach.) Ach. subsp. *epibryon*
Lecanora epibryon subsp. *broccha* (Nyl.) Lumbsch
Lecanora farinacea Fée
Lecanora galactiniza Nyl.
Lecanora mobergiana Lumbsch & Elix
Lecanora saligna (Schrad.) Zahlbr.
Lecanora wilsonii Müll.Arg.
Lecanora sp. [HO 589520]
Lecidea atromorio C.Knight
Lecidea capensis Zahlbr.
Lecidea fuscoatrula Nyl.
Lecidea ochroleuca Pers.
Lecidella flavovirens Kanvilas & Elix
Lecidella sublapicida (C.Knight) Hertel
Lecidella xylogena (Müll.Arg.) Kantvilas & Elix
Lepraria caesioalba (de Lesd.) J.R.Laundon
Lepraria finkii (B. de Lesd.) R.C.Harris
Lepraria jackii Tønsberg
Lepraria sp. [HO 590136]
Leptogium pecten F.Wilson
Leptogium victorianum F.Wilson
Megalaria grossa (Pers. ex Nyl.) Hafellner
Megalaria melaloma (C.Knight) Kantvilas
Menegazzia subpertusa P.James & D.J.Galloway
Micarea cf. *intersociella* (Stirt.) Coppins
Micarea melaneida (Nyl.) Coppins
Monerolechia badia (Fr.) Kalb
Mycocalicium victoriae (C.Knight ex F.Wilson) Tibell
Neophyllis melacarpa (F.Wilson) F.Wilson
Ochrolechia africana Vain.
Ochrolechia blandior (Nyl.) Darb.
Ochrolechia gyrophorica (A.W.Archer) A.W.Archer & Lumbsch
Pannoparmelia angustata (Pers.) Zahlbr.
Pannoparmelia wilsonii (Räsänen) D.J.Galloway
Paraporpidia leptocarpa (C.Bab. & Mitt.) Hertel & Rambold
Parmelia erumpens Kurok.
Parmeliella nigrocincta (Mont.) Müll.Arg. *s. lat.*
Parmelinopsis afrorevoluta (Krog & Swinscow) Elix & Hale
Parmotrema perlatum (Huds.) M.Choisy
Parmotrema reticulatum (Taylor) M.Choisy
Pertusaria lophocarpa Körb.
Pertusaria pertractata Stirt.
Pertusaria sp. [HO 590069]
? *Pertusaria* sp. [HO 598371]
Physcia neonubila Elix
? *Placidium* sp. [HO 589330]
Porpidia soredizodes (Lamy) Knoph, Hertel & Rambold
Pseudocyphellaria neglecta (Müll.Arg.) H.Magn.
Psora crystallifera (Taylor) Müll.Arg.
Punctelia pseudocoralloidea (Gyeln.) Elix & Kantvilas
"Ramalodium" sp. [HO 590067]
Ramboldia arandensis (Elix) Kalb, Lumbsch & Elix
Ramboldia blastidiata Kantvilas & Elix
Ramboldia brunneocarpa Kantvilas & Elix
Ramboldia laeta (Stirt.) Kalb, Lumbsch & Elix
Ramboldia petraeoides (Nyl. ex C.Bab & Mitt.) Kantvilas & Elix
Ramboldia plicatula (Müll.Arg.) Kantvilas & Elix

Appendix 1.3 cont.

Ramboldia sorediata Kalb
Ramboldia stuartii (Hampe) Kantvilas & Elix
Ramboldia subnexa (Sirt.) Kantvilas & Elix
Rhizocarpon geographicum (L.) DC.
Rhizocarpon reductum Th.Fr.
Rhizocarpon viridiatrum (Wulfen) Körb.
Rinodina asperata (Shirley) Kantvilas
Rinodina confusa H.Mayrhofer & Kantvilas
Rinodina obscura Müll.Arg.
Rinodina teniswoodiorum Elix & Kantvilas
Rinodina thiomela (Nyl.) Müll.Arg.
Schaereria sp. [HO 594256]
Schismatomma aff. *rediuntum* (Hasse) Tehler [HO 591822, 591824]
Schismatomma occultum (C.Knight & Mitt.) Zahlbr.
Tephromela alectoronica Kalb
Tephromela atra (Huds.) Hafellner
Tephromela granularis Kantvilas
Tephromela sorediata Kalb & Elix
Trapelia glebulosa (Sm.) J.R.Laundon
Trapelia lilacea Kantvilas & Elix
Trapelia sp. [HO 589519]
Trapeliopsis flexuosa (Fr.) Coppins & P.James
Trapeliopsis granulosa (Hoffm.) Lumbsch
Usnea cornuta Körb.
Usnea rubrotincta Stirt.
Usnea scabrida Taylor subsp. *scabrida*
Verrucaria tasmanica P.M.McCarthy
Xanthoparmelia amplexula (Stirt.) Elix & J.Johnst.
Xanthoparmelia digitiformis (Elix & P.Armstr.) Filson
Xanthoparmelia elixii Filson
Xanthoparmelia flavescentireagens (Gyeln.) D.J.Galoway
Xanthoparmelia microcephala Elix & Kantvilas
Xanthoparmelia mougeotina (Nyl.) D.J.Galloway
Xanthoparmelia neotinctina (Elix) Elix & J.Johnst.
Xanthoparmelia scabrosa (Taylor) Hale
Xanthoparmelia segregata Elix & J.Johnst.
Xanthoparmelia subprolixa (Nyl. ex Kremp.) O.Blanco *et al.*
Xanthoparmelia tasmanica (Hook.f. & Taylor) Hale
Xanthoparmelia verrucella (Essl.) O.Blanco *et al.*

Appendix 1.4: Invertebrate taxa of *Wind Song*

ARTHROPODS

BLATTODEA (COCKROACHES AND TERMITES)
BLABERIDAE (COCKROACHES)
Calolampra unplaced
BLATTIDAE (COCKROACHES)
Platyzosteria melanaria (Erichson, 1842)
ECTOBIIDAE (COCKROACHES)
Ectobiidae unplaced
TERMITOIDAE (TERMITES)
Termitoidae unplaced

COLEOPTERA (BEETLES)

ADERIDAE (ANT-LIKE LEAF-BEETLES)
Aderidae unplaced
BELIDAE (BELID WEEVILS)
Rhinotia bimaculata (Pascoe, 1871)
Rhinotia haemoptera Kirby, 1819
BUPRESTIDAE (JEWEL-BEETLES)
Agrilus assimilis australis Thomson, 1879
CANTHARIDAE (SOLDIER-BEETLES)
Chauliognathus lugubris (Fabricius, 1801)
Chauliognathus tricolor (Castelnau, 1840)
Heteromastix unplaced
CARABIDAE (GROUND-BEETLES)
Adelotopus unplaced
Agonocheila unplaced
Anomotarus illawarrae (Macleay, 1873)
Hypharpax peronii (Castelnau, 1867)
Notiobia quadricollis (Chaudoir, 1878)
Sarothrocrepis unplaced
Scopodes unplaced
CERAMBYCIDAE (LONGHORN-BEETLES)
Ancita crocogaster (Boisduval, 1835)
Bethelium diversicorne (White, 1846)
Cerambycidae unplaced
Stenoderus concolor W.S. Macleay, 1826
Tessaromma undatum Newman, 1840
CHRYSOMELIDAE (LEAF-BEETLES)
Arsipoda TFIC sp. 02
Cadmus crucicollis (Boisduval, 1835)
Ditropidus subaeneus Chapuis, 1875
Eboo viridula (Erichson, 1842)
Eurispa albipennis (Germar, 1848)
Monolepta cribriceps Lea, 1923
Monolepta unplaced
Paropsisterna nobilitata (Erichson, 1842)
Peltoschema De Little sp.3
Peltoschema hamadryas (Stål, 1860)
Peltoschema oceanica (Boisduval, 1835)
Peltoschema orphana (Erichson, 1842)
CLERIDAE (CHECKERED-BEETLES)
Blackburniella hilaris (Westwood, 1849)
Eleale aspera Newman, 1841
Eunatalis porcata (Fabricius, 1787)
Lemidia cicatricosa Lea, 1907
Lemidia nitens (Newman, 1841)
Lemidia subaenea Gorham, 1877
Pylus bicinctus (Newman, 1842)
COCCINELLIDAE (LADYBIRDS)
Coccinella transversalis Fabricius, 1781
Coccinella undecimpunctata Linnaeus, 1758
Harmonia conformis (Boisduval, 1835)
Rhyzobius TFIC sp. 09
Rhyzobius unplaced
CORYLOPHIDAE (MINUTE HOODED-BEETLES)
Corylophidae unplaced
CRYPTOPHAGIDAE (SILKEN FUNGUS-BEETELS)
Cryptophagidae unplaced
CURCULIONIDAE (WEEVILS)
Curculionidae unplaced
Gonipterus scutellatus Gyllenhal, 1833
Gonipterus unplaced
Melaemosaccus ocularis (Pascoe, 1873)
Merimnetes oblongus (Blanchard, 1853)
Orthorhinus klugii Boheman, 1835
Poropterus TFIC sp. 04
Scotasmus carinirostris Boheman, 1842
Scotasmus litoralis (Lea, 1911)
ELATERIDAE (CLICK-BEETLES)
Agrypnus impressicollis (Elston, 1924)
Agrypnus pictipennis (Candèze, 1857)
Conoderus basalis (Gyllenhal, 1817)
Conoderus erubescens (Candèze, 1859)
Conoderus unplaced
Elateridae unplaced TFIC sp. 10
Elateridae unplaced

Appendix 1.4 cont.

EROTYLIDAE (PLEASING FUNGUS-BEETLES)
Thallis vinula Erichson, 1842
LATRIDIIDAE (MINUTE BROWN SCAVENGER-BEETLES)
Corticariinae unplaced
LEIODIDAE (ROUND FUNGUS-BEETLES)
Zeadolopus unplaced
MELANDRYIDAE (FALSE DARKLING-BEETLES)
Orchesia minuta Lea, 1908
Orchesia TFIC sp. 01
MORDELLIDAE (PINTAILED BEETLES)
Hoshihananomia leucosticta (Germar, 1848)
Mordella TFIC sp. 04
Mordellidae unplaced TFIC sp. 04
Mordellidae unplaced
NITIDULIDAE (SAP-BEETLES)
Carpophilus TFIC sp. 02
OEDEMERIDAE (FALSE BLISTER-BEETLES)
Dohrnia miranda Newman, 1851
PHALACRIDAE (SHINING FLOWER-BEETLES)
Phalacridae unplaced TFIC sp. 05
Phalacridae unplaced
Phalacrus uniformis (Blackburn, 1891)
PTINIDAE (DEATHWATCH-BEETLES)
Deltocryptus unplaced
Lasioderma serricorne (Fabricius, 1792)
Ptinus exulans Erichson, 1842
Ptinidae unplaced
SALPINGIDAE (NARROW-WAISTED BARK-BEETLES)
Neosalpingus hybridus (Erichson, 1842)
SCARABAEIDAE (SCARAB-BEETLES)
Automolius depressus (Blanchard, 1850)
Diphucephala colaspidoides (Gyllenhal, 1817)
Heteronyx hirtuosus Blackburn, 1890
Heteronyx unplaced
Liparetrus convexus Boisduval, 1835
Onthophagus australis Guérin-Méneville, 1838
Onthophagus fuliginosus Erichson, 1842
Onthophagus pronus Erichson, 1842
SCRAPTIIDAE (FALSE FLOWER-BEETLES)
Scraptiidae unplaced
SILPHIDAE (CARRION-BEETLES)
Ptomaphila lacrymosa (Schreibers, 1802)
STAPHYLINIDAE (ROVE-BEETLES)
Aleocharinae unplaced
Oxytelinae unplaced
Paederus unplaced
Phloeocharinae unplaced
Pselaphinae unplaced
Tachyporinae unplaced
TENEBRIONIDAE (DARKLING-BEETLES)
Adelium brevicorne Blessig, 1861
Atoichus bicolor (Blackburn, 1893)
Coripera deplanata (Boisduval, 1835)
Isopteron obscurum (Erichson, 1842)
Lepispilus sulcicollis (Boisduval, 1835)
Meneristes australis (Boisduval, 1835)
Pemanoa tasmanica (Carter, 1915)
THROSCIDAE (FALSE CLICK-BEETLES)
Aulonothroscus elongatus (Bonvouloir, 1859)
TROGOSSITIDAE (BARK-GNAWING BEETLES)
Leperina decorata (Erichson, 1842)

DERMAPTERA (EARWIGS)
LABIDURIDAE
Labidura riparia (Pallas, 1773)

DIPTERA (TRUE FLIES)
ACROCERIDAE (SPIDER-FLIES)
Ogcodes flavescens White, 1914
ASILIDAE (ROBBERFLIES)
Cabasa pulchella (Macquart, 1846)
Cerdistus unplaced
Daptolestes limbipennis (Macquart, 1846)
Laphria rufifemorata Macquart, 1846
Leptogaster unplaced
Neoscleropogon unplaced
Zosteria alcetas (Walker, 1849)
BOMBYLIIDAE (BEEFLIES)
Aleucosia atherix (Newman, 1841)
Anthrax maculatus Macquart, 1846
Docidomyia puellaris White, 1916
Exechohypopion unplaced
Staurostichus unplaced
Villa fuscicostata (Macquart, 1846)
CALLIPHORIDAE (BLOWFLIES)
Calliphora stygia (Fabricius, 1782)
Calliphora unplaced
Calliphorinae unplaced
CHLOROPIDAE (FRIT-FLIES)
Gaurax unplaced
DITOMYIIDAE (DITOMYIID FUNGUS-GNATS)
Ditomyiidae unplaced
DOLICHOPODIDAE (LONG-LEGGED FLIES)
Heteropsilopus cingulipes (Walker, 1835)
Heteropsilopus ingenuus (Erichson, 1842)
Medetera unplaced
Narabeenia spinipes Bickel, 1994
Dolichopodidae unplaced
DROSOPHILIDAE (VINEGAR-FLIES)
Drosophilidae unplaced
HYBOTIDAE (DANCE-FLIES)
Hybotidae unplaced
EPHYDRIDAE (SHORE-FLIES)
Ephydridae unplaced
FANNIIDAE (LESSER HOUSEFLIES)
Fanniidae unplaced
KEROPLATIDAE (KEROPLATID FUNGUS-GNATS)
Keroplatidae unplaced
LAUXANIIDAE (LAUXANIID FLIES)
Lauxaniidae unplaced
LIMONIIDAE (LIMONIID CRANEFLIES)
Gynoplistia unplaced
MUSCIDAE (HOUSEFLIES)
Helina unplaced
Pygophora apicalis Schiner, 1868
Muscidae unplaced
NEMESTRINIDAE (TANGLE-VEINED FLIES)
* *Trichophthalma* unplaced
PIPUNCULIDAE (BIG-HEADED FLIES)
Pipunculidae unplaced
PLATYSTOMATIDAE (SIGNAL-FLIES)
Duomyia decora (Macquart, 1846)
Rivellia unplaced
RHAGIONIDAE (SNIPE-FLIES)
Atherimorpha vernalis White, 1914
Rhagionidae unplaced
SARCOPHAGIDAE (FLESH-FLIES)
Metopia nudibasis (Malloch, 1930)
STRATIOMYIDAE (SOLDIER-FLIES)
Boreoides tasmanensis Bezzi, 1922
Lecomyia caerulea (White, 1914)
Odontomyia unplaced

Appendix 1.4 cont.

SYRPHIDAE (HOVERFLIES)
Eumerus argyrogaster Ferguson, 1926
Eumerus latipes Macquart, 1846
Melangyna viridiceps (Macquart, 1847)
Psilota unplaced
TABANIDAE (MARCH-FLIES)
Dasybasis unplaced
TACHINIDAE (BRISTLE-FLIES)
Chaetophthalmus similis (Walker, 1853)
Heterometopia argentea Macquart, 1846
Rutilia unplaced
Senostoma unplaced
Trigonospila unplaced
Tachinidae unplaced
THEREVIDAE (STILETTO-FLIES)
Agapophytus quatiens (White, 1916)
Anabarhynchus unplaced
Ectinorhynchus unplaced
Evansomyia phyciformis (White, 1916)
TIPULIDAE (TIPULID CRANE-FLIES)
Tipulidae unplaced

HEMIPTERA (TRUE BUGS)
ALYDIDAE (BROAD-HEADED BUGS)
Mutusca brevicornis (Dallas, 1852)
CICADELLIDAE (CICADELLID LEAFHOPPERS)
Deltocephalinae unplaced
Ledromorpha planirostris (Donovan, 1805)
Penthimiini unplaced
Putoniessa nigra (Walker, 1862)
Rhotidus teliformis (Walker, 1851)
Tartessinae unplaced
Cicadellidae unplaced
CICADIDAE (CICADAS)
Diemeniana euronotiana (Kirkaldy, 1909)
CIXIIDAE (CIXIID LEAFHOPPERS)
Cixiinae unplaced
CLASTOPTERIDAE (CLASTOPTERID LEAFHOPPERS)
Pectinariophyes stalii (Spångberg, 1878)
CYDNIDAE (BURROWING-BUGS)
Adrisa atra (Dallas, 1851)
FLATIDAE (FLATID LEAFHOPPERS)
Siphanta tasmanica Fletcher, 1985
Siphanta cf. *hebes* (Walker, 1851)
MIRIDAE (MIRID PLANT-BUGS)
Deraeocoris unplaced
Pseudopantilius australis (Walker, 1873)
Miridae unplaced
OCHTERIDAE (SHORE-BUGS)
Ochterus unplaced
PENTATOMIDAE (STINK-BUGS)
Dictyotus caenosus (Westwood, 1837)
Diemenia rubromarginata (Guérin, 1831)
Omyta centrolineata (Westwood, 1837)
Pentatomidae unplaced
PIESMATIDAE (ASH-GREY LEAF-BUGS)
Mcateella unplaced
PSYLLIDAE (PSYLLID BUGS)
Psyllidae unplaced
REDUVIIDAE (ASSASSIN-BUGS)
Coranus trabeatus Horváth, 1902
Empicoris rubromaculatus (Blackburn, 1889)
Gminatus australis (Erichson, 1842)
Peirates fuliginosus (Erichson, 1842)
Peirates unplaced
* *Ptilocnemus* unplaced
RHYPAROCHROMIDAE (DIRT-COLOURED SEED-BUGS)
Euander lacertosus (Erichson, 1842)
SCHIZOPTERIDAE (JUMPING SOIL-BUGS)
Pateena sp. nr *polymitarior* Hill, 1980

HYMENOPTERA (ANTS, BEES AND WASPS)
AMPULICIDAE (COCKROACH-WASPS)
Ampulicidae unplaced
APIDAE (HONEYBEES AND ALLIES)
i *Apis mellifera* Linnaeus, 1758
Exoneura bicolor Smith, 1854
BETHYLIDAE (BETHYLID WASPS)
Bethylidae unplaced
BRACONIDAE (BRACONID WASPS)
Braconinae unplaced
Braconidae unplaced
CHRYSIDIDAE (CUCKOO-WASPS)
Chrysidinae unplaced
Chrysididae unplaced
COLLETIDAE (PLASTERER-BEES)
Hylaeus perhumilis (Cockerell, 1914)
CRABRONIDAE (CRABRONID WASPS)
Bembix furcata Erichson, 1842
Crabroninae unplaced
Pison unplaced
Podagritus unplaced
Sphodrotes unplaced
Tachysphex unplaced
Crabronidae unplaced
EVANIIDAE (HATCHET-WASPS)
Evaniidae unplaced
FORMICIDAE (ANTS)
* *Amblyopone australis* Erichson, 1842
Anonychomyrma biconvexa (Santschi, 1928)
Anonychomyrma nitidiceps (E. André, 1896)
* *Camponotus consobrinus* (Erichson, 1842)
Camponotus hartogi Forel, 1902
Camponotus unplaced
Hypoponera unplaced
Myrmecia forficata (Fabricius, 1787)
Myrmecia pilosula Smith, 1858
Pheidole unplaced
Polyrhachis unplaced
Rhytidoponera tasmaniensis Emery, 1898
Formicidae unplaced
GASTERUPTIIDAE (GASTERUPTIID WASPS)
Gasteruptiidae unplaced
ICHNEUMONIDAE (ICHNEUMON-WASPS)
Echthromorpha intricatoria (Fabricius, 1804)
Hyposoter unplaced
Ichneumon promissorius (Erichson, 1842)
Netelia unplaced
Ophioninae unplaced
Ichneumonidae unplaced
MUTILLIDAE (VELVET-ANTS)
Odontomyrme cordatiformis Lelej, 1983
POMPILIDAE (SPIDER-HUNTING WASPS)
Turneromyia
Ageniellini unplaced
Ctenostegus unplaced
Pompilidae unplaced
SPHECIDAE (THREAD-WAISTED WASPS)
Prionyx unplaced
TIPHIIDAE (FLOWER-WASPS)
Diamma bicolor Westwood, 1835
Tachynomyia abdominalis (Guérin-Méneville, 1842)
Thynnoides mesopleuralis Turner, 1912
VESPIDAE (POTTER-WASPS AND SOCIAL WASPS)
Australozethus tasmaniensis Giordani Soika, 1969

Appendix 1.4 cont.

Paralastor unplaced
i *Vespula germanica* (Fabricius, 1793)

LEPIDOPTERA (MOTHS AND BUTTERFLIES)
ANTHELIDAE (AUSTRALIAN LAPPET MOTHS)
Anthela cnecias Turner, 1921
Anthela repleta (Walker, 1855)
CARPOSINIDAE (FRUITWORM MOTHS)
Carposinidae unplaced
COSMOPTERIGIDAE (COSMET MOTHS)
Cosmopterigidae unplaced
Limnaecia unplaced
Macrobathra sp. Flinders CB10
Macrobathra unplaced
CRAMBIDAE (GRASS MOTHS)
Crambidae unplaced
Eudonia cleodoralis (Walker, 1859)
Eudonia unplaced
Glaucocharis ANIC sp. 10
Nacoleia rhoeoalis (Walker, 1859)
Parapoynx euryscia (Meyrick, 1885)
Ptochostola microphaeellus (Walker, 1866)
Scoparia exhibitalis Walker, 1866
Scoparia plagiotis Meyrick, 1887
ELACHISTIDAE (GRASS-MINER MOTHS)
Elachistidae unplaced
EREBIDAE
Crioa hades (Lower, 1903)
Halone sejuncta (R. Felder & Rogenhofer, 1875)
Orgyia anartoides (Walker, 1855)
Pantydia sparsa Guenée, 1852
Philenora aspectalella (Walker, 1864)
Praxis edwardsii Guenée, 1852
Praxis porphyretica Guenée, 1852
Rhapsa suscitatalis (Walker, 1859)
Sandava scitisignata (Walker, 1862)
Spilosoma glatignyi (Le Guillou, 1841)
Thallarcha jocularis (Rosenstock, 1885)
Trigonistis asthenopa Meyrick, 1902
GELECHIIDAE (TWIRLER MOTHS)
Anarsia unplaced
GEOMETRIDAE (LOOPERS, INCHWORMS)
Anachloris uncinata (Guenée, 1857)
Antasia flavicapitata (Guenée, 1857)
Arhodia lasiocamparia Guenée, 1858
Capusa senilis Walker, 1857
Casbia farinalis (Rosenstock, 1885)
Casbia melanops Rosenstock, 1885
Chloroclystis approximata (Walker, 1869)
Chloroclystis filata (Guenée, 1858)
Chlorocoma dichloraria (Guenée, 1857)
Chlorocoma externa (Walker, 1861)
Chlorocoma vertumnaria (Guenée, 1857)
Chrysolarentia leptophrica (Turner, 1922)
Circopetes obtusata (Walker, 1860)
Corula geometroides Walker, 1856
Crypsiphona ocultaria (Donovan, 1805)
Cyneoterpna wilsoni (Felder & Rogenhofer, 1875)
Dichromodes confluaria (Guenée, 1857)
Dichromodes stilbiata (Guenée, 1857)
Didymoctenia exsuperata (Walker, 1860)
Dissomorphia australiaria (Guenée, 1857)
Ectropis bispinaria (Guenée, 1857)
Ectropis despicata (Walker, 1860)
Ectropis excursaria (Guenée, 1857)
Ectropis fractaria (Guenée, 1857)
Casbia tetramera Lower, 1894
Epidesmia hypenaria (Guenée, 1857)
Epyaxa subidaria (Guenée, 1857)
Gastrina cristaria Guenée, 1857
Gastrinodes bitaeniaria (Le Guillou, 1841)
Hydriomenini unplaced nr *severata* (Guenée, 1857)
Hydriomenini unplaced *severata* (Guenée, 1857)
Idiodes apicata Guenée, 1857
Melanodes anthracitaria Guenée, 1857
Microdes diplodonta Turner, 1904
Microdes squamulata Guenée, 1858
Microdes unplaced
Nisista serrata (Walker, 1857)
Oenochroma vetustaria (Walker, 1860)
Oenochroma vinaria Guenée, 1857
Phelotis cognata (Walker, 1860)
Prasinocyma semicrocea (Walker, 1861)
Psilosticha mactaria (Guenée, 1857)
Rhinodia rostraria Guenée, 1857
Rhynchopsota delogramma Lower, 1903
Scioglyptis lyciaria (Guenée, 1857)
Scioglyptis sp. CB02
Scopula optivata (Walker, 1861)
Syneora cheleuta (Meyrick, 1892)
GLYPHIPTERIGIDAE (SEDGE MOTHS)
Glyphipteryx cometophora (Meyrick, 1880)
GRACILLARIIDAE (LEAF BLOTCH MINER MOTHS)
Gracillariidae unplaced
Polysoma eumetalla (Meyrick, 1880)
HELIOZELIDAE (SHIELD BEARER MOTHS)
Heliozelidae unplaced
HESPERIIDAE (SKIPPERS)
$ *Antipodia chaostola leucophaea* (Couchman, 1946)
Hesperilla donnysa aurantia Waterhouse, 1927
Pasma tasmanica (Miskin, 1889)
$ *Taractrocera papyria* (Boisduval, 1832)
Trapezites lutea glaucus Hübner, 1819
HYPERTROPHIDAE (TWIG MOTHS)
Eupselia carpocapsella (Walker, 1864)
Hypertrophidae unplaced
LASIOCAMPIDAE (SNOUT MOTHS)
Pararguda nasuta (Lewin, 1805)
Pararguda rufescens (Walker, 1855)
Pernattia pusilla (Donovan, 1805)
LECITHOCERIDAE (LONG-HORNED MOTHS)
Lecithocera terrigena (Meyrick, 1904)
LIMACODIDAE (CUP MOTHS)
Doratifera pinguis (Walker, 1855)
LYCAENIDAE (COPPER, HAIRSTREAK AND BLUE BUTTERFLIES)
* *Paralucia aurifer* (Blanchard, 1848)
* *Pseudalmenus chlorinda chlorinda* (Blanchard, 1848)
NOCTUIDAE (OWLET MOTHS, CUTWORMS, ARMYWORMS)
Aedia leucomelas (Linnaeus, 1758)
Agrotis munda Walker, 1857
Agrotis porphyricollis Guenée, 1852
Bathytricha truncata (Walker, 1856)
Dasygaster sp. ANIC01
Hadenini unplaced species inquirenda *dictyota* Lower, 1902
Hadenini unplaced species inquirenda *ligniplena* Walker, 1857
Leucania uda Guenée, 1852
Noctuidae unplaced
Proteuxoa bistrigula (Walker, 1857)
Proteuxoa sp. nr *flexirena* (Walker, 1865)
NOLIDAE (TUFT MOTHS)
Elesma BYRNE 'Wind Song sp. 01'
Nola pleurosema (Turner, 1944)
Nola unplaced
Uraba lugens Walker, 1863
NOTODONTIDAE (PROMINENTS)
Hobartina amblyiodes (Turner, 1931)

Appendix 1.4 cont.

Neola semiaurata Walker, 1855
Psalidostetha banksiae (Lewin, 1805)
Sorama bicolor Walker, 1855

NYMPHALIDAE (ADMIRAL AND BROWN BUTTERFLIES)
* *Geitoneura klugii* (Guérin-Méneville, 1830)
Heteronympha merope (Fabricius, 1775)
* *Junonia villida* (Fabricius, 1787)
$ *Vanessa itea* (*Fabricius,* 1775)
* *Vanessa kershawi* (McCoy, 1868)
OECOPHORIDAE (CONCEALER MOTHS)
Agriophara BYRNE 'Wind Song sp. 01
Agriophara unplaced'
Barea unplaced
Euchaetis inceptella (Walker, 1864)
Eulechria melesella (Newman, 1856)
Ocystola crystallina Meyrick, 1885
Oecophoridae unplaced
Oecophorinae genus nr *Poliorhabda* unplaced
Oecophorinae sp. 01 CB Wind Song
Oecophorinae unplaced
Oxythecta hieroglyphica Meyrick, 1885
Philobota sp. ANIC66
Philobota unplaced
Phryganeutis unplaced
Stathmopoda cephalaea Meyrick, 1897
Stathmopoda chalcotypa Meyrick, 1897
Stathmopodinae unplaced
Thalerotricha mylicella Meyrick, 1884
Zacorus unplaced
PIERIDAE (WHITE BUTTERFLIES)
*i *Pieris rapae* (Linnaeus, 1758)
PLUTELLIDAE (DIAMONDBACK MOTHS)
Plutella xylostella (Linnaeus, 1758)
PYRALIDAE (SNOUT MOTHS, GRASS MOTHS)
Ctenomeristis almella (Meyrick, 1879)
Galleriinae unplaced
Gauna aegusalis (Walker, 1859)
Phycitinae unplaced
Pyralidae unplaced
SATURNIIDAE (EMPEROR MOTHS)
Opodiphthera helena (White, 1843)
SPHINGIDAE (HAWK MOTHS)
Hippotion scrofa (Boisduval, 1832)
TINEIDAE (FUNGUS MOTHS)
Tineinae unplaced
TORTRICIDAE (LEAFROLLER MOTHS)
Acropolitis rudisana (Walker, 1863)
Anisogona mediana (Walker, 1863)
Asthenoptycha unplaced
Capua sp. AT21
Constrictana constrictana (Walker, 1866)
Epiphyas postvittana (Walker, 1863)
Epiphyas xylodes (Meyrick, 1910)
Epitymbia isoscelana (Meyrick, 1881)
Eucosmini unplaced
Euphona euphona (Meyrick, 1910)
Holocola triangulana Meyrick, 1881
Holocola unplaced
Olethreutinae unplaced
Thrincophora lignigerana (Walker, 1863)
Tortricidae unplaced
Tortricinae unplaced
YPONOMEUTIDAE (ERMINE MOTHS)
Yponomeutinae unplaced
ZYGAENIDAE (FORESTERS)
Pollanisus viridipulverulenta (Guérin-Méneville, 1839)

MANTODEA (PRAYING MANTISES)
MANTIDAE
* *Tenodera australasiae* (Leach, 1814)

MECOPTERA (HANGING-FLIES AND ALLIES)
BITTACIDAE
Harpobittacus australis (Klug, 1838)

NEUROPTERA (LACEWING AND ANTLIONS)
HEMEROBIIDAE (HEMEROBIID LACEWINGS)
Hemerobiidae unplaced
MYRMELEONTIDAE (ANTLIONS)
Myrmeleon acer Walker, 1853

ODONATA (DRAGONFLIES AND DAMSELFLIES)
LESTIDAE (SPREAD-WINGED DAMSELFLIES)
* *Austrolestes psyche* (Hagen, 1862)
LIBELLULIDAE (SKIMMER DRAGONFLIES)
* *Orthetrum caledonicum* (Brauer, 1865)

ORTHOPTERA (GRASSHOPPERS AND CRICKETS)
ACRIDIDAE (GRASSHOPPERS)
Austroicetes vulgaris (Sjöstedt, 1931)
* *Goniaea australasiae* (Leach, 1814)
Phaulacridium vittatum (Sjöstedt, 1920)
Tasmaniacris tasmaniensis (Bolívar, 1898)
Acrididae unplaced
GRYLLACRIDIDAE (RASPY-CRICKETS)
Kinemania ambulans (Erichson, 1842)
RHAPHIDOPHORIDAE (CAMEL-CRICKETS)
Parvotettix domesticus Richards, 1970
TETRIGIDAE (GROUNDHOPPERS)
Paratettix argillaceus (Erichson, 1842)
Tetrigidae unplaced
TETTIGONIIDAE (BUSH-CRICKETS OR KATYDIDS)
Caedicia simplex (Walker, 1869)
Zaprochilus australis (Brullé, 1835)
TRIGONIDIIDAE (SWORD-TAIL CRICKETS)
Bobilla unplaced
Trigonidium albovittata (Chopard, 1951)

PHASMIDA (STICK-INSECTS)
PHASMATIDAE
* *Ctenomorpha marginipennis* Gray, 1833

OTHER ARTHROPODS

ACARI (MITES)
TETRANYCHIDAE (SPIDER-MITES)
*i *Tetranychus lintearius* Dufour, 1832

ARANEAE (SPIDERS)
ARANEIDAE (ORB-WEAVING SPIDERS)
* *Dolophones conifera* (Keyserling, 1886)
* *Eriophora pustulosa* (Walckenaer, 1842)
* *Plebs bradleyi* (Keyserling, 1887)
DESIDAE (HOUSE-SPIDERS)
* *Badumna insignis* (L. Koch, 1872)
LYCOSIDAE (WOLF-SPIDERS)
* *Tasmanicosa godeffroyi* (L. Koch, 1865)
SALTICIDAE (JUMPING-SPIDERS)
* *Myrmarachne luctuosa* (L. Koch, 1879)
SPARASSIDAE (HUNTSMAN SPIDERS)
* *Delena cancerides* Walckenaer, 1837
* *Neosparassus diana* (L. Koch, 1875)
TETRAGNATHIDAE (LONG-JAWED SPIDERS)
* *Tetragnatha* unplaced
THERIDIIDAE (COMB-FOOTED SPIDERS)
* *Ariamnes patersoniensis* Hickman, 1927
* *Euryopis splendens* (Rainbow, 1916)

Appendix 1.4 cont.

THOMISIDAE (CRAB-SPIDERS)
* *Diaea* species
* *Hedana valida* L. Koch, 1875

SCORPIONES (SCORPIONS)
BOTHRIURIDAE
* *Cercophonius squama* (Gervais, 1844)

DIPLOPODA (MILLIPEDES)
JULIDAE
*i *Ommatoiulus moreleti* (Lucas, 1860)

CHILOPODA (CENTIPEDES)
SCOLOPENDRIDAE
* *Cormocephalus westwoodi* (Newport, 1844)

ISOPODA (SLATERS)
ARMADILLIDIIDAE
*i *Armadillidium vulgare* (Latreille, 1804)
PORCELLIONIDAE
*i *Porcellio scaber* Latreille, 1804

MOLLUSCA (SNAILS AND SLUGS)
RHYTIDIDAE
Tasmaphena ruga (Legrand, 1871)
Prolesophanta nelsonensis (Brazier, 1871)
CARYODIDAE
Caryodes dufresnii (Leach, 1815)
PUNCTIDAE
Paralaoma hobarti (Cox, 1868)
Paralaoma cf *morti* (Cox, 1864)
Paralaoma mucoides (Tenison-Woods, 1879)
Gratilaoma sp. "Knocklofty"
Laomavix collisi (Brazier, 1877)
Punctidae sp. "Micro Cripps" (genus undetermined)
CHAROPIDAE
Tasmathera sp. (possibly new)
ARIONIDAE
*i *Arion intermedius* Normand, 1852
HELICARIONIDAE
Helicarion cuvieri Ferussac, 1821
HELICIDAE
*i *Cornu aspersum* (Muller, 1774)
LIMACIDAE
*i *Lehmannia nyctelia* (Bourguignat, 1861)
OXYCHILIDAE
i *Oxychilus alliarius* (Miller, 1822)

OTHER INVERTEBRATES

ANNELIDA (LEECHES AND EARTHWORMS)
DOMANIBDELLIDAE
* *Philaemon grandis* Ingram, 1957

PLATYHELMINTHES (FLATWORMS)
GEOPLANIDAE
* *Anzoplana trilineata* Winsor, 2006
* *Australopacifica nichollsi* (Dendy, 1915)

Appendix 1.5: Vertebrate taxa of *Wind Song*

FROGS

* *Crinia signifera* Girard, 1853
* *Limnodynastes dumerilii insularis* Parker, 1940
* *Litoria ewingii* (Duméril & Bibron, 1841)

REPTILES

$ *Austrelaps superbus* (Günther, 1858)
* *Niveoscincus metallicus* (O'Shaughnessy, 1874)
* *Rankinia diemensis* (Gray, 1841)
* *Tiliqua nigrolutea* (Quoy & Gaimard, 1824)

MAMMALS

* *Macropus rufogriseus rufogriseus* (Desmarest, 1817)
* *Tachyglossus aculeatus setosus* (Geoffroy, 1803)
* *Thylogale billardierii* (Desmarest, 1822)
* *Trichosurus vulpecula fuliginosus* (Ogilby, 1831)
* *Vombatus ursinus tasmaniensis* (Spencer & Kershaw, 1910)

BIRDS

$ *Acanthiza chrysorrhoa* (Quoy & Gaimard, 1830)
* *Acanthiza pusilla diemenensis* Gould, 1838
$ *Acanthorhynchus tenuirostris* (*Latham,* 1801)
$ *Anthochaera chrysoptera* (Latham, 1801)
$ *Anthus richardi* Vieillot, 1818
i* *Alauda arvensis Linnaeus,* 1758
* *Anas castanea* (Eyton, 1838)
* *Anthochaera chrysoptera tasmanica* (Mathews, 1912)
* *Aquila audax* (Latham, 1801)
* *Artamus cyanopterus cyanopterus* (Latham, 1801)
* *Cacomantis flabelliformis* (Latham, 1801)
* *Cacomantis pallidus* (Latham, 1801)
* *Calyptorhynchus funereus xanthanotus* Gould, 1838
i* *Carduelis carduelis* (*Linnaeus,* 1758)
* *Chrysococcyx basalis* (Horsfield, 1821)
* *Chrysococcyx lucidus plagosus* (Latham, 1801)
* *Colluricincla harmonica harmonica* (Latham, 1801)
* *Coracina n. novaehollandiae* (Gmelin, 1789)
* *Corvus tasmanicus* Mathews, 1912
* *Cracticus tibicen hypoleuca* Gould, 1837
* *Cracticus torquatus cinereus* (Gould, 1837)
i* *Dacelo novaeguineae novaeguineae* (Hermann, 1783)
* *Egretta novaehollandiae* (Latham, 1790)
* *Epthianura albifrons* (Jardine & Selby, 1828)
* *Falco berigora tasmanica* (Mathews, 1916)
$ *Glossopsitta concinna* (Shaw, 1791)
$ *Haliaeetus leucogaster* Gmelin, 1788
* *Hirundo neoxena* (Gmelin, 1789)
* *Lathamus discolor* (Shaw, 1790)
* *Malurus cyaneus cyaneus* (Ellis, 1782)
* *Manorina melanocephala melanocephala* (Latham, 1801)
$ *Melithreptus affinis* (Lesson, 1839)
$ *Myiagra cyanoleuca* (Vieillot, 1818)
* *Ninox novaeseelandiae leucopsis* (Gould, 1838)
* *Pardalotus punctatus punctatus* (Shaw, 1792)
* *Pardalotus striatus striatus* (Gmelin, 1789)
* *Petrochelidon nigricans nigricans* (Vieillot, 1817)
* *Petroica boodang leggii* (Sharpe, 1879)
* *Petroica phoenicea* Gould, 1837
* *Phalacrocorax carbo novaehollandiae* Stephens, 1826
* *Phaps chalcoptera* (Latham, 1790)
$ *Phylidonyris novaehollandiae* (Latham, 1790)
* *Platycercus caledonicus* (Gmelin, 1788)
* *Platycercus eximius* (Shaw, 1792)
* *Podargus strigoides strigoides* (Latham, 1801)
* *Rhipidura albiscapa albiscapa* Gould, 1840
* *Strepera versicolor arguta* Gould, 1846
i* *Sturnus vulgaris* Linnaeus, 1758
i* *Turdus merula merula* Linnaeus, 1758
$ *Vanellus miles* (Boddaert, 1783)
$ *Zosterops lateralis* (Latham, 1801)

INSECT SUCCESSION PATTERN ON DECOMPOSING PIG CARCASSES IN TASMANIA: A SUMMER STUDY

by Paola A. Magni, J. David North, Melle Zwerver, Ian R. Dadour

(with one text-figure, two plates and one table)

Magni, P.A., North, J.D., Zwerver, M., Dadour, I.R. 2019 (14:xii). Insect succession pattern on decomposing pig carcasses in Tasmania: a summer study. *Papers and Proceedings of the Royal Society of Tasmania* **153:** 31–38. ISSN 0080–4703. Discipline of Medical, Molecular & Forensic Sciences, Murdoch University, 90 South St, Murdoch, Western Australia 6150, Australia (PAM, IRD). Murdoch University Singapore, King's Centre, 390 Havelock Road, Singapore 169662 (current address: PAM). Tasmania Police Forensic Services, 37–43 Liverpool Street, Hobart, Tasmania 7000, Australia (JDN). Tasmania Police, Devonport Police Station, 24 Wenvoe Street, Devonport, Tasmania 7310, Australia (MZ). *Author for correspondence: Email: P.Magni@murdoch.edu.au.

Insect succession has been studied around the world using the predictable and mostly sequential arrival pattern of different insect species that are attracted to a decomposing carcass. In cases of suspicious death of humans and animals, carrion insects may be used to assist in crime scene reconstruction. The present research represents the first study in forensic entomology to be undertaken in Tasmania, investigating insect succession patterns on decomposing pig carcasses and providing a preliminary database of forensically important insects. Six pig carcasses were placed in two contrasting locations (rural and urban) in northern Tasmania. Insect successional waves were recorded over a 40-day study during the austral summer season. Results showed that decomposition rates and insect assemblages varied between each location. Eleven insect taxa, representing nine families, were identified in association with the decomposition of the pig carcasses at both localities. Blowflies present on the pig carcasses throughout the decomposition process were *Calliphora stygia* Malloch (Diptera: Calliphoridae) at both sites and *Lucilia sericata* (Meigen) at the urban site only. These preliminary results will provide useful information in any future casework involving human remains and associated insect material in Tasmania.

Key Words: decomposition, Tasmania, insect succession, forensic entomology, blowflies.

INTRODUCTION

Forensic entomology is the study of insects and other arthropods associated with legal investigations (Voss *et al.* 2009). Forensic entomology is a method used worldwide to assist in determining a more accurate time since death or *minimum Post Mortem Interval* (minPMI) (Goff 1993, Voss *et al.* 2009). Other data gleaned from insect succession patterns have become invaluable during legal investigations. They can be used in conjunction with other methods to assist in the overall crime scene reconstruction, movement of the body, the presence of toxicological substances (Wallace 2017, Magni *et al.* 2018) and foreign DNA in the alimentary tract (Carvahlo *et al.* 2005, Di Luise *et al.* 2008). Insects can also indicate the presence of lesions in cases of suspicious death (Byrd & Castner 2010).

The decomposition of organic matter involves a very complex biological process whereby the body undergoes several different — but generally predictable — stages (typically fresh, bloated, advanced, dry, skeletal) (Goff 1993). The decomposition process is largely dependent on factors such as geographical location and environmental conditions (habitat, biogeography and accessibility by the fauna), deposition conditions (e.g., surface, burial or submersion) and climatic conditions (e.g., temperature, season) (Voss *et al.* 2011, Hyde *et al.* 2015, Roberts *et al.* 2017). Furthermore, extrinsic and intrinsic properties that each human cadaver exhibits — e.g., gut microbiome, size, cause of death, presence/absence of clothes — should also be considered (Gill 2005, Voss *et al.* 2009, Benbow *et al.* 2015, Iqbal *et al.* 2018).

During the process of decomposition, remains can attract a variety of organisms that utilise the cadaver as a food source, a reproduction site, and as a predator and/or parasite on the fauna present on the remains (Payne 1965, Schotsmans *et al.* 2017). In general, the majority of the fauna present on the remains is represented by insects, mainly as blowflies, flesh flies and house flies (Family Diptera) and carrion beetles (Family Coleoptera) (Morris & Dadour 2015). The arrival of insects is not random, but associated with certain stages of decomposition (Mégnin 1894). Insect succession involves the process of invasion by insects onto the carcass soon after death and continues by attracting subsequent insect species in successional waves (Smith 1986, Gill 2005, Voss *et al.* 2008).

Several studies have shown that approximately 48–72 hours after a death, the use of insects attracted to the body is the most accurate method to estimate the minPMI (Goff 1991, Byrd & Castner 2010, Marks *et al.* 2010). To provide reliable information for investigation purposes an entomologist must correctly identify the insects' species and age, and if possible utilise an appropriate database of the insect succession pattern for the specific geographical region where the remains were discovered. In Australia, the majority of insect succession studies that have been published are based in New South Wales and the Australian Capital Territory (Fuller 1934, Johnson *et al.* 2013, Forbes *et al.* 2014, Gherlenda *et al.* 2016, Barton *et al.* 2017), Victoria (Archer & Elgar 2003, Archer *et al.* 2005), Queensland (O'Flynn 1983, Farrell *et al.* 2015) and Western Australia (Bornemissza 1957, Dadour *et al.* 2001,

O'Brien *et al.* 2007, Voss *et al.* 2008, 2009, 2011, Magni *et al.* 2019). To date, there has been one published study detailing insect succession patterns on decomposing remains in Tasmania (Lang *et al.* 2006). That study concerned the species of blowflies that emerge from a possum carcass and how they might affect blowfly strike of sheep over autumn and winter. No emphasis was placed on how such species might be used in a forensic context. Tasmania has been geographically and genetically isolated for long periods (McCalman 2009), and as a consequence entomological data on necrophagous insects from this region rather than mainland Australia are essential in legal investigations to assist in determining a minPMI.

The aim of the present study is to provide a preliminary database, detailing forensically important insects, mainly blowfly species, attracted to decomposing carcasses at two locations in northern Tasmania during the austral summer. Commonalities and dissimilarities were investigated at two contrasting habitats, *rural* and *urban* over a 40-day study period.

MATERIALS AND METHODS

Study sites

A 40-day survey of animal decomposition was conducted at two study sites from 12 December 2014 to 20 January 2015. The first study site (*rural site*, RS) was located within a paddock situated approximately 8 km northeast of Devonport, Tasmania (41°10'26.6"S, 146°17'23.6"E) (pl. 1C, D). The site was adjacent to an area used primarily for potato cultivation. No pesticides were used during the 40-day trial. The second study site (*urban site*, US) was located within a scientific facility (Department of Primary Industries, Parks, Water and Environment Facility) situated approximately 5 km south of Launceston, Tasmania (41°28'09.8"S, 147°08'28.6"E) (pl. 1A, B). The area used for the study was approximately 0.13 km south of the main facility: it was surrounded by a grass paddock with buildings, roads and trees nearby. The two study sites were situated approximately 100 km apart.

PLATE 1 — Study locations, urban site (A, B) and rural site (C, D). Credit for A and C: Google maps https://www.google.com/maps/.

Experimental design

In Australia and in many other countries, the field-based research of surface decomposition of human remains is legally prohibited outside of licensed facilities such as the Australian Facility for Taphonomic Experimental Research (Smith 1986, Knobel *et al.* 2018, Luong *et al.* 2018). As a result, animal models, such as dogs, rats, pigs and guinea pigs as well as bait and sticky-based fly traps are the only option available for research relating to insect succession data and to provide information based on geographic location and seasonality pertaining to minPMI (Dadour *et al.* 2001). Pigs (*Sus scrofa domesticus* Erxleben) are the preferred animal model and most commonly used (Schoenly & Hall 2002), as they represent the most reliable facsimile to humans. Furthermore, pigs are reasonably inexpensive and can be readily available in numbers with uniform mass and size similar to humans (Dadour *et al.* 2001, Gill 2005). In the present study, a total of six pigs weighing 45 ± 5 kg were used. The pigs were obtained from a commercial piggery and within two hours of being head bolted, the pig carcasses were transported and placed in each of the study sites. This represented Day 1 of the experiment. In both the RS and US, three pigs were placed 10 m apart from each other and protected by individual scavenger-proof cages constructed by the authors (Dadour *et al.* 2001). The design of the mesh (metal) cage prevented animal scavenging but still allowed insect access.

Sampling of the insect fauna was conducted over a 40-day period. Representative samples of insect fauna were collected in accordance with forensic entomology guidelines. Insects colonising the carcasses were sampled daily for the first week and subsequently every two to four days for the 40-day period. Following collection, samples were labelled in relation to collection site, sampling date and time, carcass number and anatomical site of collection. As per the best practice in forensic entomology (Amendt *et al.* 2007), half the insect samples were sacrificed to allow the identification of the age at the time of collection, while the other half were allowed to reach the adult instar stage to facilitate identification (Byrd & Tomberlin 2010). For this purpose, fauna boxes 25×14×13 cm with a base of white sand and beef liver placed on top were used to rear the immatures (Byrd & Tomberlin 2010).

The preserved samples and trapped adult fly samples were observed under a steromicroscope (Leica® MZ8). Larvae were aged in terms of instar and taxonomically identified to family/genus/species) by an insect taxonomist. The preserved samples were separated and organised according to collection site, carcass number and date of collection. Due to its forensic perspective, this study was mostly focused on blowflies.

Environmental data

Daily records of ambient temperature were obtained from weather stations at the sites of decomposition and surrounds from local weather stations in both localities (fig. 1). The weather stations were within 5 km from the RS and US sites respectively. Records of rainfall levels were obtained from the Bureau of Meteorology website (Bureau of Meteorology 2017) (fig. 1).

Assessment of the stage of decomposition

In addition to insect sampling, all carcass replicates were observed and assessed daily for the first week and subsequently every two to four days for the 40-day period. Photographs of the carcasses were taken each observation day (pl. 2). The decomposition process was determined using the five stages described by Goff (1993). Carcasses were considered in the fresh stage from the moment of death to the onset of bloat. Bloat commenced when inflation of the carcass occurred and concluded upon deflation of the carcass. The decay stage was considered from carcass deflation and the outer layer of the skin on the carcass becomes broken leaving only dry constituents remaining which is the commencement of the dry stage. Carcasses were considered as skeletonised when the absence of soft tissue was noted on bones, and only cartilage and hair remained.

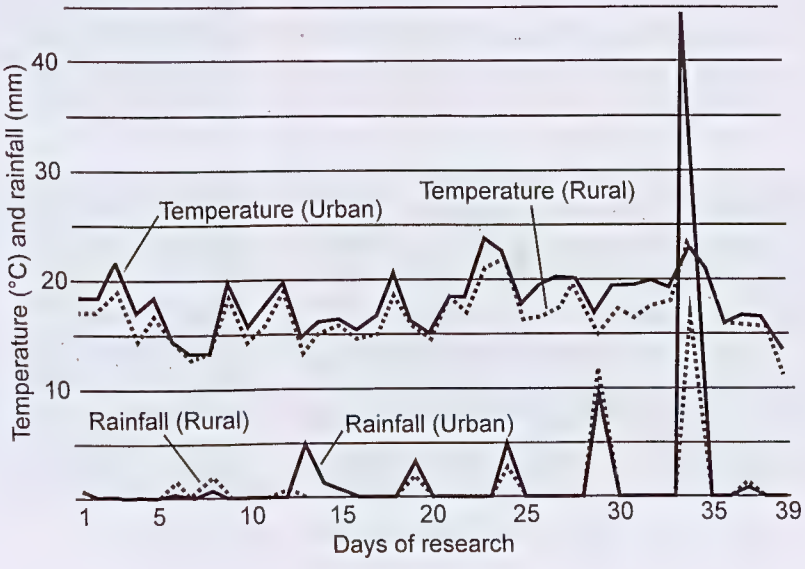


FIG. 1 – Comparative view of the environmental data (average ambient temperature and rainfall) of the two location sites, rural and urban, over the duration of the 40-day study.

URBAN SITE

RURAL SITE

Day 2 – Fresh stage
Day 2 – Fresh stage
Day 6 – Bloat stage
Day 6 – Bloat stage
Day 9 – Decay stage
Day 9 – Bloat stage
Day 11 – Dry stage
Day 27 – Dry stage
Day 29 – Skeletonization
Day 35 – Skeletonization

PLATE 2 — Decomposition process at various stages at the two study sites, urban and rural.

TABLE 1 — Summer succession of insects on carcasses at two study sites, *urban site* and *rural site*, over 40 days in Tasmania, Australia. Data are summarised by decomposition stages. Data for the three carcass repetitions were combined in this table.

URBAN SITE			Days and stage of decay																	
			1–2		3–7			8–10				11				12–40				
Order	Family	Species	Fresh		Bloat			Decay				Dry				Skeletonisation				
			E[1]	L	E	L	A	L	P	PF	A	L	P	PF	A	L	P	PF	A	
Diptera	Calliphoridae	*Calliphora stygia*	×	×		×		×				×								
		Lucilia sericata						×								×				
	Muscidae	*Musca vetustissima*										×				×				
Coleoptera	Dermestidae	spp.														×			×	
	Cleridae	spp.														×				
	Histeridae	spp.																	×	

RURAL SITE			Days and stage of decay																	
			1–3		4–9			10–24				25–31				32–40				
Order	Family	Species	Fresh		Bloat			Decay				Dry				Skeletonisation				
			E	L	E	L	A	L	P	PF	A	L	P	PF	A	L	P	PF	A	
Diptera	Calliphoridae	*Calliphora stygia*	×	×	×	×		×	×	×			×				×			
	Muscidae	*Musca vetustissima*				×		×		×		×		×		×		×		
Coleoptera	Forficulidae	spp.																	×	
	Staphylinidae	spp.					×	×				×				×				
	Dermestidae	spp.					×					×			×	×			×	
	Cleridae	spp.									×				×				×	
	Silphidae	spp.				×	×	×							×					
	Carabidae	spp.									×									
	Histeridae	spp.					×				×									

[1] Life stages are identified by E (eggs), L (larvae), P (pupae), PF (larva in post-feeding instar) and A (adults).
× = specimen collection.

RESULTS

Decomposition process and environmental data

The progression and duration of each decomposition stage were not entirely comparable between the two sites over the 40 days (table 1). While the first stages of decomposition were approximately the same duration (Fresh stage: US 3 days, RS 3 days; Bloat stage: US 4 days, RS 5 days), the subsequent stages required a different time to be completed (Decay stage: US 2 days, RS 14 days; Dry stage: US 3 days, RS 6 days; Skeletonisation stage: US from day 12 of the experiment, RS from day 32 of the experiment) (table 1). Overall, the decomposition in the RS happened at a slower rate compared to that at US. In particular, while both carcasses remained relatively fresh until Day 3, the duration of the decay stages lasted up to 24 days at RS but only 10 days at US, with all carcass replicates reaching the dry/remains stage by Day 25 (table 1, pl. 2). The reason for this may be a consequence of the environmental temperatures at the RS which typically had lower minimum temperatures than the US (fig. 1). The RS showed an average minimum temperature of 11.2°C and maximum temperature of 23.6°C, in comparison to the US site where the ambient temperatures were at an average minimum temperature of 13.2°C and maximum temperature of 22.5°C (fig. 1).

Insect succession

Throughout this study, 11 insect taxa, representing nine families, were identified in association with the decomposition of the pig carcasses (table 1). In both US and RS, representatives of the order Diptera were the primary colonisers of all the carcasses. Two genera of fly were present on the pig carcasses throughout the decomposition process at both the RS and US. Early colonisers arriving during the fresh stage of decomposition were *Calliphora stygia* Malloch (Diptera: Calliphoridae). The secondary coloniser, *Lucilia sericata* (Meigen) (Diptera: Calliphoridae) reached the pig carcasses during the bloat stage, but only in the US. A review of the biology and distribution of *Lucilia* species is detailed by Zumpt (1965), and the *Calliphora* by Wallman & Adams (1997). Additional colonisers arriving later during decomposition (at 10 days) included the flies of the family Muscidae (*Musca vetustissima* Walker) (Hughes 1970) (pl. 2).

Eggs and larvae of *C. stygia* were observed on carcasses at US from Day 1 of the experiment, and were present up until the skeletonisation stage. *L. sericata*, instead, appeared on Day 8, while *M. vetustissima* and various carrion beetles were more prevalent after Day 11. In contrast, eggs and larvae of *C. stygia* were observed on the pig carcasses present at the RS as well, but only up until the dry/remains stage. Pupae of *C. stygia* were then present until the skeletonistion stage of decomposition. *M. vetustissima* was present during the early stages of decay and remained present until the skeletonisation stages. Beetles were also present at various stages, but in no predictable order during the decomposition process.

DISCUSSION

This is the first forensically focused study recording the decomposition process and the succession patterns of necrophagous insects on decomposing pig carcasses at two locations in Tasmania during the austral summer.

Comparable research that may be considered relevent was previously conducted some 450 km away on mainland Australia in Victoria (Archer & Elgar 2003). The Victorian study highlighted that insect succession patterns change according to the geographical region, but as the study did not account for temperatures, they were only able to suggest that seasonal differences were probably important. In accordance with this research, the present study shows that both the decomposition process and the insect assemblages collected from the carcasses placed in two different environmental locations, RS and US, differed between and over the study period (table 1). As only one previous study has been published in Tasmania during the autumn/winter period (Lang *et al.* 2006), a comparative analysis was unable to be conducted to determine whether the insect assemblages observed at the two locations are consistent throughout Tasmania.

The different environmental temperatures recorded at the two sites were found to be the main factors that affected both the duration and the extent of the decomposition of the carcasses. The environmental temperatures at the RS were typically lower in comparison to the US, leading to a slower rate of decomposition at the RS and a difference in insect colonisation (table 1). In both US and RS, insects species belonging to the family Calliphoridae appeared early on in the decomposition process stages and remained for the 40 days of the study. In comparison, the *M. vetustissima* family appeared at a much later stage (from Day 11) of the decomposition process and remained present for the rest of the study. This pattern of insect succession is consistent with previous studies within Australia, even when the study was carried out for longer periods (Voss *et al.* 2008, 2009, McIntosh *et al.* 2017). However, these studies contained a much larger array of insect colonisers as they were recorded over one or two seasons in different habitat types and geographical regions.

The validity of applying collected reference data from one habitat for analysis in another within the same geographic region is considered precarious in forensic cases when estimating the time since death based on insect assemblages (Abd El-bar & Sawaby 2011, Alves *et al.* 2014). Variation between habitats is known to alter decomposition rates and thus influence insect succession patterns (Campobasso *et al.* 2001). As evident in this study, decomposition rates vary according to different habitat types (pl. 2) as well as variations in insect colonisation, but, for the most part, the insect succession patterns were reasonably consistent (table 1). Variations in insect succession patterns are usually related to the species' habitat preferences (Voss *et al.* 2009). Although present at the US site, *L. sericata* was not detected at the RS site. One explanation is that *Lucilia* species have a preference for higher temperatures compared to *Calliphora* species (Smith 1986). As well, *L. sericata* tends to prefer urban habitats only in Australia (Monzu 1977). Carrion beetles were not compared in this study as a part of the insect succession patterns because the beetles are more of an opportunistic insect that come and utilise remains irregularly over time (Smith 1986). In fact, the study of Barton *et al.* (2017) highlights this factor that predaceous beetles are more prevalent early on coinciding with high numbers of fly larvae, and beetle scavengers are present late in the decomposition stages. The review on dermestids (beetle scavengers) by Magni *et al.* (2015) contrasts these results showing that these beetles occur from three days to three years following death.

The results from this study — although the first with a forensic focus in Tasmania — will provide fundamental baseline reference data on fly succession on a carcass. As such, ongoing research in the area of insect succession patterns (flies and beetles) needs to be expanded to include seasonal and habitat types within every geographic region.

At present, a limited but expanding number of studies have been focused on insect succession in Australia (Morris & Dadour 2015). For example, insect succession patterns have not yet been investigated in most of northern Australia and this becomes a limiting factor in any forensic case conducted there. Furthermore, continuing studies are also required in Tasmania during different seasons, habitat types and should include an array of different circumstantial situations, such as clothed vs unclothed, buried vs exposed, and concealed vs exposed carcasses (Voss *et al.* 2009).

ACKNOWLEDGEMENTS

The authors would like to thank C. K. & S. Farquhar for their support in supplying the pigs for the study and the active rural paddock near Devonport. The Department of Primary Industries, Parks, Water and Environment Facility is thanked for allowing the use of their land in Launceston as the urban decomposition site for the study. We thank Ms Tracey Fong for her contribution to the literature review and the two reviewers for their helpful comments.

REFERENCES

Abd El-bar, M.M. & Sawaby, R.F. 2011: A preliminary investigation of insect colonization and succession on remains of rabbits treated with an organophosphate insecticide in El-Qalyubiya Governorate of Egypt. *Forensic Science International* **208**: 26–30.

Alves, A.C., Santos, W.E., Farias, M.L. & Creao-Duarte, A.J. 2014: Blowflies (Diptera, Calliphoridae) associated with pig carcasses in a Caatinga area, Northeastern Brazil. *Neotropical Entomology* **43**: 122–126.

Amendt, J., Campobasso, C.P., Gaudry, E., Reiter, C., LeBlanc, H.N. & Hall, M. 2007. Best practice in forensic entomology. Standards and guidelines. *International Journal of Legal Medicine* **121**: 104–109.

Archer, M.S. & Elgar, M.A. 2003: Yearly activity patterns in southern Victoria (Australia) of seasonally active carrion insects. *Forensic Science International* **132**: 173–176.

Archer, M.S., Bassed, R.B., Briggs, C.A. & Lynch, M.J. 2005: Social isolation and delayed discovery of bodies in houses: the value of forensic pathology, anthropology, odontology and entomology in the medico-legal investigation. *Forensic Science International* **151**: 259–265.

Barton, P.S., Evans, M.J., Pechal, J.L. & Benbow, M.E. 2017: Necrophilous insect dynamics at small vertebrate carrion in a temperate eucalypt woodland. *Journal of Medical Entomology* **54**: 964–973.

Benbow, M.E., Pechal, J.L., Lang, J.M., Erb, R. & Wallace, J.R. 2015. The potential of high-throughput metagenomic sequencing of aquatic bacterial communities to estimate the Postmortem Submersion Interval. *Journal of Forensic Science* **60**: 1500–1510

Bornemissza, G. 1957: An analysis of arthropod succession in carrion and the effect of its decomposition on the soil fauna. *Australian Journal of Zoology* **5**: 1–12.

Byrd, J.H. & Castner, J.L. 2010: *Forensic Entomology – The Utility of Arthropods in Legal Investigation* (Second Edition). CRC Press, Boca Raton, USA: 681 pp.

Byrd, J.H. & Tomberlin J.K. 2010: Laboratory rearing of forensic insects. *In* Byrd, J.H. & Castner, J.L. (eds): *Forensic Entomology – The Utility of Arthropods in Legal Investigation* (Second Edition) CRC Press, Boca Raton, USA: 177–200.

Bureau of Meteorology 2017: Climate statistics for Australian locations. Commonwealth of Australia, Australia.

Campobasso, C.P., Di Vella, G. & Introna, F. 2001: Factors affecting decomposition and Diptera colonization. *Forensic Science International* **120**: 18–27.

Carvahlo, F., Dadour, I.R., Groth, D.M. & Harvey, M.L. 2005: Isolation and detection of ingested DNA from the immature stages of *Calliphora dubia* (Diptera: Calliphoridae): a forensically important blowfly. *Forensic Science, Medicine, and Pathology* **1**: 261–265.

Dadour, I.R., Cook, D.F., Fissioli, J.N. & Bailey, W.J. 2001: Forensic entomology: application, education and research in Western Australia. *Forensic Science International* **120**: 48–52.

Di Luise, E., Magni, P.A., Staiti, N., Spitaleri, S. & Romano, C. 2008: Genotyping of human nuclear DNA recovered from the gut of fly larvae. *Forensic Science International: Genetics Supplement Series* **1**: 591–592.

Farrell, J.F., Whittington, A.E. & Zalucki, M.P. 2015: A review of necrophagous insects colonising human and animal cadavers in south-east Queensland, Australia. *Forensic Science International* **257**: 149–154.

Forbes, S.L., Perrault, K.A., Stefanuto, P.H., Nizio, K.D. & Focant, J.F. 2014: Comparison of the decomposition VOC profile during winter and summer in a moist, mid-latitude (Cfb) climate. *PLoS ONE* 9: e113681.

Fuller, M. 1934: The insect inhabitants of carrion: A study in animal ecology. *CSIRO Bulletin* **9**: 5–63.

Gherlenda, A.N., Esveld, J.L., Hall, A.A., Duursma, R.A. & Riegler, M. 2016: Boom and bust: rapid feedback responses between insect outbreak dynamics and canopy leaf area impacted by rainfall and CO_2. *Global Change Biology* **22**: 3632–3641.

Gill, G.J. 2005: Decomposition and Arthropod Succession on Above Ground Pig Carrion in Rural Manitoba. Unpublished PhD thesis, University of Manitoba, Manitoba.

Goff, M.L. 1991: Comparison of insect species associated with decomposing remains recovered inside dwellings and outdoors on the island of Oahu, Hawaii. *Journal of Forensic Science* **36**: 748–753.

Goff, M.L. 1993: Estimation of postmortem interval using arthropod development and successional patterns. *Forensic Science Review* **5**: 81–94.

Hughes, R.D. 1970: The seasonal distribution of bushfly (*Musca vetustissima* Walker) in south-east Australia. *Journal of Animal Ecology* **39**: 691–706.

Hyde, E.R., Haarmann, D.P., Petrosino, J.F., Lynne, A.M. & Bucheli, S.R. 2015: Initial insights into bacterial succession during human decomposition. *International Journal of Legal Medicine* **129**: 661–671.

Iqbal, M.A., Ueland, M. & Forbes, S.L. 2018: Recent advances in the estimation of post-mortem interval in forensic taphonomy. *Australian Journal of Forensic Science* DOI: 10.1080/00450618.2018.1459840

Johnson, A.P., Mikac, K.M. & Wallman, J.F. 2013: Thermogenesis in decomposing carcasses. *Forensic Science International* **231**: 271–277.

Knobel, Z., Ueland, M., Nizio, K.D., Patel, D. & Forbes, S.L. 2018: A comparison of human and pig decomposition rates and odour profiles in an Australian environment. *Australian Journal of Forensic Science* **51**: 557–572.

Lang, M., Allen, G. & Horton, B.P. 2006: Blowfly succession from possum (*Trichosurus vulpecula*) carrion in a sheep-farming zone. *Medical and Veterinary Entomology* **20**: 445–452.

Luong, S., Forbes, S.L., Wallman, J.F. & Roberts, R.G. 2018. Monitoring the extent of vertical and lateral movement of human decomposition products through sediment using cholesterol as a biomarker. *Forensic Science International* **285**: 93–104.

Magni, P.A., Petersen, C., Georgy, J. & Dadour, I.R. 2019: The effect of suitcase concealment on insect colonization: a pilot study in Western Australia. *Global Journal of Forensic Science & Medicine* **1**: 1–9.

Magni, P.A., Pazzi, M., Droghi, J., Vincenti, M. & Dadour, I.R. 2018: Development and validation of an HPLC-MS/MS method for the detection of ketamine in *Calliphora vomitoria* (L.) (Diptera: Calliphoridae). *Journal of Forensic and Legal Medicine* **58**: 64–71.

Magni, P.A., Voss, S.C., Testi, R., Borrini, M. & Dadour, I.R. 2015: A biological and procedural review of forensically significant dermestes species (Coleoptera: Dermestidae). *Journal of Medical Entomology* **52**: 755–769.

Marks, M.K., Love, J.C. & Dadour, I.R. 2010: Taphonomy and time: estimating the postmortem interval. *In* D. Steadman (ed.), *Hard Evidence: Case Studies in Forensic Anthropology* (Second Edition). CRC Press: pp. 165–178.

McCalman, I., 2009: *Darwin's Armada.* Penguin Books: 423 pp.

McIntosh, C.S., Dadour, I.R. & Voss, S.C. 2017: A comparison of carcass decomposition and associated insect succession onto burnt and unburnt pig carcasses. *International Journal of Legal Medicine* **131**: 835–845.

Mégnin, J.P. 1894: *La faune de cadavres. Application de l'entomologie à la médicine légale.* Encyclopedie Scientifique des Aide-Mémoire. G. Masson, Paris: 214 pp.

Monzu, N. 1977: Coexistence of Carrion Breeding Calliphoridae (Diptera) in Western Australia. Unpublished PhD thesis, University of Western Australia.

Morris, B. & Dadour, I.R. 2015: Insects and their uses in legal cases. *In* Freckleton, I. & Selby, H. (eds): *Expert Evidence.* The Law Book Company Limited: P8-5291-5298-5381.

O'Brien, R.C., Forbes, S.L., Meyer, J. & Dadour, I.R. 2007: A preliminary investigation into the scavenging activity on pig carcasses in Western Australia. *Forensic Science, Medicine and Pathology* **3**: 194–199.

O'Flynn, M.A. 1983: The succession and rate of development of blowflies in carrion in southern Queensland and the application of these data to forensic entomology. *Journal of the Australian Entomological Society* **22**: 137–148.

Payne, J.A. 1965: A summer carrion study of the baby pig *Sus scrofa* Linnaeus. *Ecology* **46**: 592–602.

Roberts, L.G., Spencer, J.R. & Dabbs, G.R. 2017: The effect of body mass on outdoor adult human decomposition. *Journal of Forensic Science* **62**:1145–1150

Schoenly, K.G. & Hall, R.D. 2002: *Testing Reliability of Animal Models in Research and Training Programs in Forensic Entomology.* NIJ Report: 34 pp.

Schotsmans, E.M., Forbes, S.L. & Márquez-Grant, **N.** 2017: *Taphonomy of Human Remains: Forensic Analysis of the Dead and the Depositional Environment.* John Wiley & Sons: 452 pp.

Smith, K.G.V. 1986: *A Manual of Forensic Entomology.* Trustees of the British Museum, London: 205 pp.

Voss, S.C., Forbes, S.L. & Dadour, I.R. 2008: Decomposition and insect succession on cadavers inside a vehicle environment. *Forensic Science, Medicine, and Pathology* **4**: 22–32.

Voss, S.C., Spafford, H. & Dadour, I.R. 2009: Annual and seasonal patterns of insect succession on decomposing remains at two locations in Western Australia. *Forensic Science International* **193**: 26–36.

Voss, S.C., Cook, D.F. & Dadour, I.R. 2011: Decomposition and insect succession of clothed and unclothed carcasses in Western Australia. *Forensic Science International* **211**: 67–75.

Wallace, D.R. 2017: Evolution of forensic entomotoxicology. *Toxicololgy and Forensic Medicine Open Journal*: SE(1) Se1–Se4, https://doi: 10.17140/TFMOJ-SE-1-e001.

Wallman, J.F. & Adams, M.D. 1997: Molecular systematics of Australian carrion-breeding blowflies of the genus Calliphora (Diptera:Calliphoridae). *Australian Journal of Zoology* **45**: 337–356.

Zumpt, F. 1965: *Myiasis in Man and Animals in the Old World.* Butterworths, London: 267 pp.

(Accepted 15 August 2019)

TAXONOMIC AND CONSERVATION STATUS OF *DOCKRILLIA STRIOLATA* (RCHB.F.) RAUSCHERT (ORCHIDACEAE) IN TASMANIA

by Mark Wapstra

(with five text-figures, three plates, two tables and one appendix)

Wapstra, M. 2019 (14:xii). Taxonomic and conservation status of *Dockrillia striolata* (Rchb.f.) Rauschert (Orchidaceae) in Tasmania. *Papers and Proceedings of the Royal Society of Tasmania* 153: 39–51. ISSN 0080-4703. Environmental Consulting Options Tasmania, Lenah Valley, Tasmania 7008, Australia. Email: mark@ecotas.com.au

Dockrillia striolata (Rchb.f.) Rauschert, known as the yellow rock-orchid, occurs in New South Wales, Victoria and along the east coast of Tasmania. Within Tasmania, populations occur on the islands of the Furneaux Group and on mainland Tasmania (Kelvedon Hills south of Swansea through to wukalina/Mount William in the northeast). The taxonomy of the species is revised, with subsp. *chrysantha* no longer recognised as distinct based on a morphological examination of flower size and other characters. The species is restricted to Devonian granite-type rocks and Jurassic dolerite, usually in near-coastal areas and in various vegetation types. A review of the conservation status indicates that the species does not qualify as threatened under the Tasmanian *Threatened Species Protection Act 1995*, with it being well-reserved throughout its range, an extent of occurrence of ca. 7500 km², represented by at least 30 well-defined subpopulations, no apparent historical or contemporary declines in any population parameters, nor any identifiable risks.

Key Words: *Dendrobium striolatum*, *Dockrillia striolata*, Orchidaceae, rock-orchid, taxonomy, distribution, habitat, conservation.

INTRODUCTION

Most of Tasmania's approximately 214 species of native orchid (de Salas & Baker 2019) are terrestrial. Unlike sub-tropical and tropical parts of the globe that support a high diversity of epiphytic species, southern Tasmania supports only one true epiphytic orchid species, *Sarcochilus australis* (Lind.) Rchb.f. Amongst the terrestrial species, there is also one species that is a true lithophyte, currently referred to as *Dockrillia striolata* (Rchb.f.) Rauschert (de Salas & Baker 2019).

Until the mid-1990s, Tasmania's "rock-orchid" was widely known as *Dendrobium striolatum* Rchb.f., with no infrataxa recognised. *Dockrillia* Brieger was described in 1981 (Brieger 1981) as a "natural group of orchids which have been segregated from the large genus *Dendrobium*" (Clements & Jones 1996, Jones 1998). The segregate genus was accepted in more recent taxonomic treatments (e.g., Jones 1998) and incorporated into most formal and informal floras (e.g., Jones *et al.* 1999, Jones 2006). However, some broader taxonomic treatments (e.g., WCSP 2019), accepts *Dendrobium* Sw. and places *Dockrillia* Brieger into synonymy. The present paper accepts *Dockrillia* Brieger, pending a broader review of Australian orchid taxonomy.

As part of a taxonomic review of Tasmanian Orchidaceae, Jones (1998) described a new infrataxon of *Dockrillia striolata*, namely the new endemic subspecies *chrysantha* D.L.Jones, which resulted in the autonym subsp. *striolata*. The subspecies were separated on four morphological features viz. "ab subsp. *striolata* floribus parum majoribus, vivide flavis; tepalis vittis paucis vel nullis; et labello valdius crenulato, differt". That is, the new subspecies tends to have slightly larger flowers, brighter yellow tepals (petals and sepals) that lack stripes or are only faintly striped, and a labellum with more strongly crenulate margins. The two subspecies were originally also separated geographically, with subsp. *chrysantha* endemic to mainland Tasmania, and subsp. *striolata* absent from mainland Tasmania but present on the eastern Bass Strait islands and extending to Victoria and New South Wales (Jones 1998, Jones *et al.* 1999). This geographic separation was always considered minor because the gap between the northeasternmost Tasmanian mainland sites (e.g., Mount William National Park area) and Cape Barren Island was effectively no greater than disjunctions on mainland Tasmania.

There have been no formal published accounts of *Dockrillia striolata* in Tasmania. The present paper aims to document the distribution and habitat of the species in Tasmania, clarify its taxonomic and nomenclatural status, and provide information on its reservation and conservation status.

METHODS

Database and collection review

Five sources of records of native plants were reviewed to produce a complete list of all known locations of *D. striolata* in Tasmania, as follows: (1) collections at the Tasmanian Herbarium, Tasmanian Museum & Art Gallery (HO); (2) collections at the Queen Victoria Museum (QVM); (3) Department of Primary Industries, Parks, Water & Environment's *Natural Values Atlas* database (NVA, DPIPWE 2019); (4) *Atlas of Living Australia* (ALA 2019); and (5) the Australasian Virtual Herbarium (AVH 2019). The data were "cleaned" to produce a definitive worksheet of known locations of the species. "Cleaning" included removal of obvious database duplicates; removal of records lacking sufficient information to precisely place the site (e.g., records

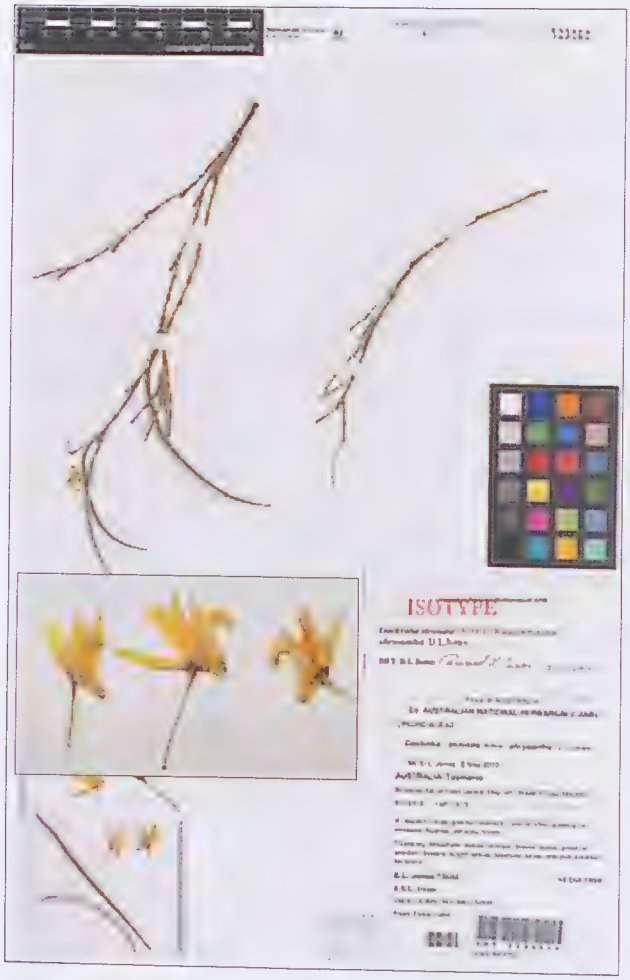


PLATE 1 — Isotype (HO523462) of *Dockrillia striolata* subsp. *chrysantha* from Bicheno, east coast of Tasmania, 18 Oct. 1994 (D.L. Jones & B.E. Jones), showing three separated flowers, all with a moderate degree of striping on the abaxial surface of the tepals [main image courtesy of the Tasmanian Herbarium; inset of three separated flowers by M. Wapstra (same flowers as in main image)].

labelled "Tasmania" or "East Coast"); and shifting of point locations to more precise sites where sufficient information was provided (e.g., records currently shown in the sea were shifted to a nearby terrestrial location if collection notes indicated an obvious location).

As it is virtually impossible to mis-identify *D. striolata* in Tasmania (it is a mono-specific genus in Tasmania), even when not in flower or fruit, because of its distinctive terete fleshy leaves and lithophytic habit, database records of the species in the NVA and ALA were taken as correct identifications to species level. Due to the taxonomic history of the species, however, collections and database records of the species had variable allocation of specimens/records to subspecific level, with many either missing this more detailed information or the subspecies clearly being allocated solely on a geographic basis (i.e. mainland Tasmanian records as subsp. *chrysantha* and Furneaux Group records as subsp. *striolata*). However, a reasonable number of specimens held at HO for mainland Tasmania had been allocated to subsp. *striolata*.

Earlier collecting localities and inclusion of the taxon in earlier treatments in floras is examined.

Comparison of records to selected variables

The "cleaned" data files were manipulated in a GIS platform (ArcGIS v. 10.5.1) to compare the point locations with the following variables (all publicly available as GIS files and/or possible to interrogate directly on The LIST, www.thelist.tas.gov.au): land tenure, with particular reference to gazetted reserves under the Tasmanian *Nature Conservation Act 2002*; geology; and elevation (by reference to the 10 m a.s.l. contour interval).

Morphometrics

All collections held at HO were examined for specimens with well-pressed flowers. Any flowers showing the complete length of a tepal were measured from the visible base of the flower to the tip of the longest tepal using Vernier™ digital calipers (precise to 1/100th mm). Appendix 1 provides a complete listing of all specimens examined.

One-way analysis of variance (ANOVA) was used to compare the length of flowers for: (A) ISLAND groups (mainland Tasmania; Furneaux islands); (B) REGIONAL groups (mainland Tasmania – east coast; mainland Tasmania – northeast; Furneaux islands); and (C) SITE groups (from south to north – Freycinet area, Bicheno/Apsley River area, St Helens area, inland northeast (Lanka Road), Mt Cameron area (including Second Sugarloaf and Lanka Road); Mt William area (including Bayleys Hill); Furneaux islands – Mt Munro; Furneaux islands – Strzelecki; Furneaux islands – Patriarchs; Furneaux islands – Killiecrankie). Tukeys (HSD) test was used to denote means that were different at the 95% confidence interval. Some sites have low sample sizes, limiting the confidence of the ANOVA between sites.

A simple linear regression was calculated to predict flower size based on latitude (with latitude allocated in decimal degrees to samples from the following sites (south to north): Freycinet area (42.14737 S); Bicheno area (41.87471 S); St Helens area (41.18693 S); Mt Cameron area (40.93421 S); inland northeast – Lanka Road (41.03957 S); Second Sugarloaf (40.98063 S); Bayleys Hill (40.97866 S); Mt Cameron area (40.9342 S1); Mt William (40.90683 S); Furneaux – Mt Munro (40.37081 S); Furneaux – Strzelecki area (40.22103 S); Furneaux – Patriarchs (39.95719 S); Furneaux – Killiecrankie (39.80903 S).

RESULTS AND DISCUSSION

Early collection history in Tasmania

The first formal collection of *Dockrillia striolata* from Tasmania was by Joseph Milligan on 10 October 1845, shown on databases as without a specific location, but is in fact the type collection of *Dendrobium milliganii* F.Muell. (a synonym of *Dockrillia striolata*), labelled "Flinders Peaks,

granite rocks" (held at MEL & BM). Milligan was appointed as the superintendent and medical officer at Wybalenna, the Aboriginal community, on Flinders Island in late 1843, and he collected widely on both Flinders Island and the smaller Furneaux islands until April 1846 (Buchanan 1988). Milligan again collected the species on 4 October 1847 from "Mt Killiecrankie, nearly opposite Sisters Isles", when he was stationed on Flinders Island for five months up to October 1847 (Buchanan 1988).

The first formal collection of the species from mainland Tasmania was in 1879 from "ridges near Ruby Plain, Georges Bay" by G.C. Smith (specimen AD 97708873). In 1880, there was a collection from the general St Helens (Georges Bay) area by an unknown collector (HO 4445), which was closely followed by collections from the same area by Leonard Rodway in 1890 and 1896. Oddly, there are no further collections or database records from the St Helens area. Later records are all from further afield such as the Mount Pearson area (and further north), but these were not until the early 1970s. It is likely that all early collections from "Georges Bay" refer to this much wider area, as evidenced by the Smith collection in 1879, which is from several kilometres west of St Helens.

FIG. 1 — Distribution of *Dockrillia striolata* in Australia [source: ALA (2019)].

Inclusions in earlier flora treatments

The species was not included in *Flora Tasmaniae* (Hooker 1858), essentially the first complete treatment of the flora of Tasmania. The omission is surprising given that the earlier collections of the species were by Milligan, one of the early colonial collectors mentioned specifically by Hooker, who cites numerous collections by him for other species, including many orchids. In *Fragmenta Phytographiæ Australiæ* (Mueller 1859), the species is treated as *Dendrobium milliganii*, with Mueller specifically recognising Milligan in his description of the species ("...legit amicus Dr. Josephus Milligan, floræ Diemenicæ indigator assiduous". Mueller (1859) makes reference to specific locations and habitats in Tasmania (viz. "in fissuris rupium graniticarum montis Strzelecki's Peak altitudine 3000' et in montibus humilioribus orientalibus insulæ Flinders' Island freti Bassii; nec non in montibus graniticis circiter 1200' altis pæninsulae Freycenetii Tasmaniæ").

It was later included in *A Handbook of the Plants of Tasmania* (Spicer 1878), suggesting the species was well-known by then. In *The Tasmanian Flora* (Rodway 1903), the species is included with distribution notes indicating "East Coast, Bass Straits; also in New South Wales and Victoria", again confirming the then-recognised distribution of the species, which remains accurate to this day. Firth (1965) in his *Native Orchids of Tasmania* included Cape Barren and Clarke islands as known locations outside mainland Tasmania. His observations are not supported by database records or collections, with only a single collection of the species from Cape Barren Island (from 1988) and none from Clarke Island. In *The Student's Flora of Tasmania Part 4A* (Curtis 1979), the species' distribution was described as "...local, especially on granite rocks at the east coast, also on Flinders Island and other islands of the Furneaux Group", this latter observation again pre-dating any formal recognition of the species from islands other than Flinders Island.

Contemporary distribution

Dockrillia striolata occurs along the Great Dividing Range from just west of Newcastle through to northeastern Victoria west of Mallacoota (fig. 1), and along the east coast of Tasmania (fig. 1).

Within Tasmania, the species is widespread in the eastern half of the State (fig. 2), occurring in the Flinders, Ben Lomond and South East bioregions. The southernmost confirmed site is in forested hills behind Kelvedon Beach, about 10 km south of Swansea. Heading northwards, the species has a somewhat disjunct distribution with clusters of records on the Freycinet Peninsula, Douglas–Apsley–Bicheno area, greater St Helens–Bay of Fires area, Mount Cameron, and Mount William areas. The northernmost record on mainland Tasmania is on Mount William itself. The species extends to Cape Barren Island and Flinders Island.

It is somewhat surprising that within the Furneaux Group of eastern Bass Strait islands the species is apparently restricted to Flinders Island and Cape Barren Island, with only one reported site on the latter (fig. 2). While many of the inner and outer Furneaux islands are low-lying and exposed to strong maritime influences, suitable substrates are available and many of the islands (such as Babel, Prime Seal, Big Dog, West Sister and Outer Sister islands; S. Harris pers. comm.) have locally suitable pockets of sheltered habitat. A comprehensive survey of the flora of the outer islands of the Furneaux Group (i.e. those excluding Flinders, Cape Barren and Clarke islands) is reported in Harris *et al.* (2001), where notably the species is not recorded. In particular, the absence of the species from the Kent Group (Deal,

FIG. 2 — Distribution of *Dockrillia striolata* in Tasmania – records from all data sources combined. The dubious outliers on the Gordon River (western circle) and at Murdunna (eastern circle) are highlighted (refer to text).

FIG. 3 — Distribution of *Dockrillia striolata* in Tasmania – records from *Natural Values Atlas* only.

FIG. 4 — Distribution of *Dockrillia striolata* in Tasmania – records from collections at the Tasmanian Herbarium (filled triangles) and the Queen Victoria Museum (open circles) only – note that the QVM collections add an important range infill at Sloop Lagoon, north of St Helens (arrowed).

FIG. 5 — Distribution of *Dockrillia striolata* in Tasmania – records from the Atlas of Living Australia (filled triangles) and the Australasian Virtual Herbarium (open circles) only. The dubious outliers on the Gordon River (western circle) and at Murdunna (eastern circle) are highlighted (refer to text).

Dover and Erith islands) about halfway between Flinders Island and Wilsons Promontory (Harris & Davis 1995, Kirkpatrick 1995), Rodondo Island (just off the southern tip of Wilsons Promontory) and Wilsons Promontory itself, is a noticeable data gap between northeastern Tasmania (mainland Tasmania and Flinders Island) through to the northeastern part of Victoria (fig. 1). Interestingly, Mueller (1859, p. 88) referred to the absence of the species from the superficially ideal Wilsons Promontory, viz. "stirps forsitan in tractu altiore granitico promontorii Wilsonii haud frustra quærenda erit" (i.e. an in vain search of perhaps a quarter of the granitic Wilsons Promontory). Firth (1965) reported the species from Clarke Island, which is a relatively large granite-based island between mainland Tasmania and Cape Barren Island. Interestingly, recent biological surveys have not confirmed the species from the island (NCHD 2014).

Reconciliation of database sources

Interpretation of database/collection information has shown several inconsistencies between the available data sources used in this project. Reliable "clean" data is critical to any true understanding of the distribution, management requirements and conservation status of a species. Inconsistency between databases, often created by infrequent and/or partial one-directional exchanges, is a potential source of misunderstanding of the distribution of a species. It is useful to compare the four data sources used as part of the present project to highlight key differences.

In summary, the NVA holds 116 records of *D. striolata* (fig. 3), only 33 of which are cross-held in the ALA. Of the 116 NVA records, 32 represent collections held by the Tasmanian Herbarium (HO), four collections held by the Queen Victoria Museum & Art Gallery (QVM); refer to figure 4 for distribution of records from HO and QVM, and the balance from various data sources. The ALA (fig. 5) only holds 47 records (43 discrete collections) of *D. striolata* from Tasmania, 33 of which are cross-held in the NVA. However, 14 of the ALA records are not cross-held in the NVA, importantly including several earlier records (from 1911, 1914, 1931, and 1933). The AVH (fig. 5) only holds nine records (seven discrete collections) of *D. striolata* from Tasmania, only one of which is cross-held in the NVA (i.e. the AVH includes six "missing" sites). These discrepancies between data sources highlight the need for careful interpretation of records, and ideally, a centralised system of storing all records.

In the case of *D. striolata*, examination of only formal collections held at the Tasmanian Herbarium (32) provides a reasonable picture of the distribution of the species within Tasmania (fig. 4). However, the NVA provides a much more complete representation of the species' distribution (fig. 3). Scrutiny of the ALA/AVH highlights the inconsistency between data sources (fig. 5), with those databases showing two obvious outliers (described below) not included in the NVA, and missing several records, a key one being the one from Cape Barren Island (also described below). In turn, the NVA is missing several important records held in the ALA, most notably the earliest record of the species from mainland Tasmania (G.C. Smith, AD 97708873).

The ALA is missing the record of the species from Cape Barren Island, which was cited in Jones (1998), shown on the distribution map in Jones *et al.* (1999), and which is represented by a collection at the Tasmanian Herbarium (Collier 3574), and included in the NVA.

The ALA also suggests that the species occurs on the Forestier Peninsula, putatively near Murdunna Hill (fig. 5), based on an observation included in the NVA (F. Duncan, 5 or 6 June 1983, project code "FT:dry" referring to extensive State-wide dry forest vegetation surveys). The original observer confirmed no field activities on the Forestier Peninsula in that period and suggested that the record more likely referred to a site near Mount Cameron in the northeast, with the northing entered incorrectly into the database as the easting coincides with that part of the state, where extensive fieldwork was undertaken in dry vegetation types (F. Duncan pers. comm.). This record is now marked as "bad" in the NVA (W. Potts pers. comm.).

There is an ALA database record labelled "Gordon River" (fig. 5) from 15 October 1911 by A.L. Rogers. The record is supported by a specimen (AD 97708874) held at the State Herbarium of South Australia (pl. 2). This record represents a significant outlier (fig. 5) and may also be from atypical habitat, i.e. non-coastal riparian rainforest

PLATE 2 — Specimen (AD97708874) of *Dockrillia striolata*, with inset showing clearly written label stating "Gordon River, Tasm." [images courtesy of the Board of the Botanic Gardens and State Herbarium (Adelaide, South Australia)].

vegetation. However, there is only one Gordon River in Tasmania, which runs from the King William Range south and west to Macquarie Harbour on the state's west coast. While it is tempting to discount this record as erroneous (e.g., a mix-up of locations by the original collector or perhaps a reference to a different Gordon River such as the one in Queensland), the collection sheet (with very clearly written location label) and supporting card catalogue record are identical (J. Kellermann pers. comm.). In addition, the Australasian Virtual Herbarium (AVH 2019) indicates 15 collections by A.L. Rogers, all of which are orchids, nine from Tasmania on 25 September 1907, 1 October 1911, 15 October 1911, 10 November 1911 and 13 December 1911, providing strong evidence for the specimen of *D. striolata* having at least been collected from Tasmania. It is presumed that A.L. Rogers was related to R.S. Rogers (1861–1942), widely recognised as one of South Australia's (indeed Australia's) foremost orchidologists (Gibberd 1988), and who collected widely, including in Tasmania. The "problem" with the record is that most of the Gordon River is presently in remote wilderness, a context even stronger in 1911. However, at this juncture, it is impossible to more precisely place the collection.

Habitat

Dockrillia striolata is an obligate lithophyte. Its two primary substrates in Tasmania are Devonian granite-granodiorite (and related igneous rock types) and Jurassic dolerite. Jones *et al.* (1999) stated that the species occurs occasionally on sandstone, but this is not borne out by database/collection information. While several records are associated with substrates other than granite or dolerite, these invariably have low precision and more precise placement of the records associates them with granite or dolerite. Mainland Australian records (as per the ALA) indicate numerous records on sandstone and similar substrates, suggesting that substrate type *per se* is not a limiting factor in the distribution of the species.

In Tasmania, the species tends to grow on massive rock outcrops taking advantage of crevices and imperfections in the rock surface. Smaller boulders and outcrops, especially of granite in more open forests, can also be occupied. While Jones *et al.* (1999; p. 143) suggested that "most colonies are found on east-facing sites directly exposed to moisture-laden sea breezes", this is not supported by field observations that indicate the species can occupy all parts of a rock outcrop, often including aspects sheltered from the effects of the sea. This is particularly the case for sites further inland, well away from any maritime influence.

Most Tasmanian sites of the species are within 2–10 km of the coast, the majority within the 2–6 km range. A cluster of sites in the Mount Cameron area is ca. 12–14 km from the nearest coast. The record from Mount Nicholas is also ca. 14 km from the coast. The most inland of Tasmanian sites is at Moorina Hill, ca. 27 km and 30 km from the north and east coast, respectively. Mainland Australian records (as per the ALA) indicate numerous records greater than 50 km inland (with one at least 110 km inland), suggesting that a maritime influence *per se* is not a limiting factor in the distribution of the species.

The vegetation associated with *D. striolata* in Tasmania is highly variable ranging from dry sclerophyll forests and woodlands dominated by species such as *Eucalyptus amygdalina* Labill., *E. viminalis* Labill, *E. tenuiramis* Miq. and *E. sieberi* L.A.S.Johnson to wet sclerophyll forest dominated by *E. regnans* F.Muell. The species is most strongly connected to the drier end of the vegetation spectrum although it is often associated with sheltered and naturally fire-protected sites such as gullies.

D. striolata occurs over a wide range of elevations. On mainland Tasmania, most sites are below ca. 200 m a.s.l., although several sites are at higher elevations (e.g., Mount Cameron ca. 550 m a.s.l., Mount Nicholas ca. 800 m a.s.l., The Hazards to ca. 485 m a.s.l., Bedggood Hill ca. 380 m a.s.l.). Most of the sites on Flinders Island and Cape Barren Island are below 500 m a.s.l.

Morphometrics

Following the formal description of subsp. *chrysantha* by Jones (1998), with ongoing field observations and collections it became apparent that the described differences between subsp. *chrysantha* and subsp. *striolata* were difficult to distinguish in both fresh and curated material, with obvious continuities within some of the features. Examination of publicly available images of the species from across its natural range in Australia reveals a wide range in flower size, crenulation of the labellum margin and flower colour. Images of representative flowers from across the Tasmanian geographic range of the species are provided in plate 3. While variation in several features is present, these are considered to be well within the likely expression of minor genetic differences of disjunct populations.

One-way ANOVA indicated that flower size differed significantly between mainland Tasmanian populations and Furneaux Group population [ISLAND group: R^2 = 0.3991, Adjusted R^2 = 0.3951, $F(1,148)$ = 98.307, MSE = 356.53, $p < 0.0001$], with flowers smaller on mainland Tasmania compared to the Furneaux islands (table 1). There was no significant difference between east coast and northeast coast populations on mainland Tasmania, but the Furneaux population was significantly different from both mainland Tasmanian populations [REGION group: R^2 = 0.3991, Adjusted R^2 = 0.3961, $F(2,147)$ = 49.87, MSE = 180.55, $p < 0.0001$], again with flowers smaller on mainland Tasmania compared to the Furneaux islands (table 1).

One-way ANOVA indicated that flower size differed significantly between sites [SITE group: R^2 = 0.5788, Adjusted R^2 = 0.5518, $F(9,140)$ = 49.87, MSE = 57.451, $p < 0.0001$], with populations from Freycinet, Bicheno, St Helens, inland northeast, Mt Cameron, Mt William and Killiecrankie not significantly different, all with smaller flowers (table 2). Mean flower size from the Mt Munro population is significantly smaller than that of the Strzelecki population but not to the preceding mainland Tasmanian

PLATE 3 — Examples of *Dockrillia striolata* from Tasmanian sites. (A & C) – Whalers Lookout, Bicheno, 14 Oct. 2015 (essentially the type location of subsp. *chrysantha*): note the moderate obvious striping on the abaxial surface of the tepals as well as faint striping on the adaxial surface of the tepals. (B & D) – Strzelecki walking track, Flinders Island, 3 Oct. 2017: note the similar degree of striping on the abaxial surface of the tepals and the very faint striping on the adaxial surface of the tepals.

TABLE 1 — Flower size differences between Furneaux islands and mainland Tasmanian populations

Region	Mean (mm)	SD	n	Minimum (mm)	Maximum (mm)
Furneaux	19.30	2.43	30	14.12	23.89
Mainland Tasmania (all)	15.44	1.75	120	11.24	19.42
East coast	15.20	1.93	48	11.24	19.11
Northeast	15.60	1.62	72	12.30	19.42

and Killiecrankie (northernmost Furneaux site) populations. The Strzelecki population has statistically the largest mean flower size (table 2) of all populations.

The results of the regression indicated latitude explained 27.4% of the variance ($R^2 = 0.2738$, $F(1, 148) = 55.8$, $p < 0.0001$), with flower size decreasing by 1.8474 mm for each degree of latitude. That is, the size of the flowers increases from south to north, although the Killiecrankie population (northernmost site) is highlighted as having a mean flower size which is the same as the Freycinet population (southernmost site), but the sample size for the former population is very small (table 2).

It is noted that the gradation in flower size from south to north (smaller to larger) is opposite to the description of subsp. *chrysantha*, which described mainland Tasmanian flowers as larger than those from the eastern Bass Strait islands and mainland Australia (Jones 1998). Flower size alone is too variable to be useful for separating mainland Tasmanian populations from those from mainland Australia and the Furneaux islands, with differences most likely to

TABLE 2 — Flower size between all sites (ordered north to south)

Region	Mean (mm)	SD	n	Minimum (mm)	Maximum (mm)
Furneaux (Killiecrankie)	14.33	0.29	2	14.12	14.53
Furneaux (Patriarch)	18.50	0.69	5	17.75	19.46
Furneaux (Strzelecki)	20.72	1.67	18	17.56	23.89
Furneaux (Munro)	16.96	1.16	5	16.09	18.38
Mt Cameron area	15.95	1.59	45	12.30	19.42
Mt William area	15.01	1.62	15	12.40	18.19
Inland northeast	14.20	0.21	2	14.05	14.35
St Helens area	15.21	1.54	10	12.91	17.71
Bicheno area	15.78	2.04	28	11.24	19.11
Freycinet area	14.33	1.44	20	11.85	13.51

be caused by minor genetic or phenotypic variation at the sub-population level.

The original description by Jones (1998) states that flower colour is taxonomically important for separating *D. striolata* subsp. *striolata* from subsp. *chrysantha*. If apparent minor variations in flower colour and size are accepted as unreliable characters on which to separate infrataxa (or indeed higher level taxa), and the degree of crenulation in the margin of the labellum is accepted as a subjective feature only (possibly only related to flower size), this leaves only the degree of striping on the tepals that can be reliably used to separate subsp. *chrysantha* from subsp. *striolata*. The often-cited (e.g., Jones *et al.* 1999) feature of subsp. *striolata* as having tepals with "dark stripes, particularly prominent on the outside of the perianth segments" and of subsp. *chrysantha* as having tepals that "lack stripes or are only faintly striped" fails to hold true. The images of the two subspecies provided with the type description (Jones 1998; p. 215) shows the prominently striped outside of the tepals of subsp. *striolata* from Genoa Peak in Victoria but does not show the inside of the tepals, and only properly shows the inside of the tepals of subsp. *chrysantha* from Bicheno in Tasmania (type location; pl. 1). The outside of the flowers from the latter site is mainly obscured but partial dark red stripes and reddish apices of the tepals are visible in some flowers. My own examination of fresh flowers from the type location of subsp. *chrysantha* at Bicheno (both Whalers Lookout and Lookout Rock) indicate that while the inside of the tepals may only be faintly striped, the outside of the flowers is moderately striped with dark red bands (pl. 3). Examination of online images of mainland Australian *striolata* clearly shows a wide range of striping on the inside (and outside) of the tepals, indicating a continuum in this character, insufficient to warrant subspecific (or even varietal) rank. It is considered that differences in flower colour and degree of adaxial/abaxial striping on perianth segments (pl. 3) most likely reflect minor genetic variation at the sub-population level, the differences presumably persistent because of periods of genetic isolation.

Taxonomy

The revised taxonomy is as follows:

Dockrillia striolata (Rchb.f.) Rauschert, *Feddes Repert.* 94(7-8): 447 (1983)

Dendrobium striolatum Rchb.f., *Hamburger Garten-Blumenzeitung* 13: 313 (1857); *Callista striolata* (Rchb.f.) Kuntze, *Revis. Gen. Pl.* 2: 655 (1891); *Dockrillia striolata* (Rchb.f.) Rauschert subsp. *striolata*.

TYPE: "Im Garten des Herrn Consul Schiller Von Herrn Stange cultivirt." cult., *Schiller* (holo W, *fide* Clements 1989).

Dendrobium milliganii F.Muell. *Fragm. (Mueller)* 1(4): 88, t. VI (1859).

TYPE: Flinders Peaks, granite rocks, 10 Oct. 1845, *J. Milligan 923* (lecto MEL, *fide* Clements 1989).

Dockrillia striolata (Rchb.f.) Rauschert subsp. *chrysantha* D.L.Jones, *Austral. Orchid Res.* 3: 9 (1998).

TYPE: Tasmania, Bicheno, 18 Oct. 1994, *D.L.Jones 13582 & B.E.Jones* (holo CANB).

The vernacular "yellow rock-orchid" is suggested to reflect the flower colour and lithophytic habit, in line with Jones *et al.* (1999), Jones (2006) and Wapstra *et al.* (2005).

As part of the present project, all specimens held at HO have been re-determined by the author to *Dockrillia striolata* (Rchb.f.) Rauschert, with no subspecies recognised.

Conservation status: population parameters

The Scientific Advisory Committee, established under the provisions of the TSPA, produced a set of "*Guidelines for Eligibility for Listing under the* Threatened Species Protection Act 1995" (DPIW 2008). These *Guidelines* include definitions of some key population variables that are used in most measures of conservation status, and these are explored below for *D. striolata*. These are further explored in the section on the conservation status of the species.

D. striolata is likely to be an under-collected species because of its fleshy leaves making curation difficult (the leaves and flowers also readily disarticulate, resulting in poorly presented specimens). In addition, non-threatened

species tend to be under-represented in databases. While it is recognised that there are numerous additional sites of *D. striolata* in Tasmania not represented by formal herbarium collections and/or database records, sufficient information is available to estimate several population parameters.

Extent of occurrence

A minimum convex polygon created around the known records/collections (excluding the dubious records from Murdunna and the Gordon River) equates to an extent of occurrence of ca. 7500 km^2, which is ca. 270 km and 50 km in a north–south and an east–west direction, respectively. It is highly unlikely that there have been any material changes to the extent of occurrence historically (e.g., from habitat loss). It is reasonable to assume that there will be future minor range infills and possibly limited range extensions.

Number of subpopulations or locations

Defining the number of subpopulations or locations of *D. striolata* in the traditional sense of the terms is problematic. For example, while it is simple enough to define a subpopulation of the species that occurs on Thirty Acre Creek south of Swansea because it is represented by a single database record, defining the number of subpopulations that are present on Freycinet Peninsula is impractical. There are only sixteen database records for the entire Freycinet Peninsula, two of which are obvious duplicates. Only seven of the remaining fourteen records have any location information, none better than "Coles Bay", "Freycinet Peninsula" or "Wineglass Bay Track". However, to field botanists, it is well-known that the species occurs on perhaps hundreds of rock outcrops across The Hazards and in surrounding lower-lying terrain. That said, it is interesting to note the lack of any records south of The Hazards – that is, the species is apparently absent from the southern part of the peninsula, including Schouten Island, despite superficially identical habitat to the northern part of the peninsula.

As such, using available information to estimate the number of subpopulations (or locations) is impractical, beyond defining some of the broader locations (table 1), and noting that several database records have not been considered due to lack of any supporting notes as to location.

The species is represented at six locations on the Furneaux islands: five on Flinders Island (Mount Killiecrankie, possibly the Wingaroo area (most likely Mount Blyth or Mount Boyes, both low-lying granite rises), North Patriarch, South Patriarch, and several sites in the Strzelecki range), and one on Cape Barren Island (Mount Munro). This is likely to represent a significant under-estimate of the number of locations supporting the species across the greater Furneaux region, with several sites being prospective such as Mount Kerford on Cape Barren Island (S. Harris pers. comm.). On mainland Tasmania, the species is represented at ca. 27 locations (excluding the dubious records from Murdunna and the Gordon River), the precise number is very much dependent on how a location is defined (i.e. the distance between locations, especially taking into account imprecision in database/collection information). The total number of locations is ca. 33, although there are several caveats to this (previously discussed) that indicate the number of locations is likely to be much greater.

There is no evidence of an historical decrease in the number of locations supporting the species. Given the reasonable likelihood of range infillings, there is likely to be a minor increase in the number of locations supporting the species.

Area of occupancy

It is difficult to estimate the area of occupancy because most records are not accompanied by detailed collection notes. Most sites are represented by locally dense "colonies" that in themselves occupy a few square metres. Even with hundreds, perhaps low to mid thousands of "colonies", within the species' distribution, the total area of occupancy is unlikely to exceed a few hectares at most. As with the extent of occurrence, there is no evidence that there has been an historical decrease, or that there is a reasonable anticipation of a future decrease, in the area of occupancy of the species. The concept of area of occupancy has limited applicability to this type of colony-forming species, with localised clumps scattered over severally closely positioned rock outcrops, themselves scattered widely across a whole mountain range.

Number of mature individuals

Estimating the total State-wide abundance of mature individuals for *D. striolata* is difficult due to the lack of information associated with most records and the growth habitat of the species. Some authors (e.g., Jones *et al.* 1999) refer to "colonies" of the species, referring to locally dense clumps of the species on rock outcrops, which may represent one or many individuals. However, at any particular site (e.g., Wineglass Bay lookout on Freycinet Peninsula or Whalers Lookout at Bicheno), the location supporting the species may only be represented by a small number of formal records but it is locally common, occurring at numerous sites, each of these represented by one to many "colonies". On this basis, it is only possible to make vague estimates of population abundance, herein suggested to be in the order of low to high thousands of "colonies".

Severely fragmented

The *Guidelines* describe "severely fragmented" as the situation in which increased extinction risk to the taxon results from the fact that most of its individuals are found in small and relatively isolated subpopulations, which may go extinct with a low probability of recolonisation. The *Guidelines* consider fragmentation relevant in relation to flora if it increases the risk of disease or weed invasion by increasing edge effects, increases inbreeding effects, or decreases reproductive output. The distribution of *D. striolata* is somewhat disjunct but hardly properly fragmented. In fact, it is more likely that some of the apparent gaps in the distribution (e.g., in the Douglas–Apsley area and the St Helens area) will be filled with more comprehensive reporting of the species.

Conservation status: reservation status

Dockrillia striolata is a well-reserved species with populations reported from the following gazetted formal reserves: Freycinet National Park, Coles Bay Conservation Area, Lookout Rock State Reserve, Whalers Lookout Conservation Area, Douglas-Apsley National Park, Lower Marsh Creek Regional Reserve, St Patricks Head State Reserve, Huntsmans Cap Regional Reserve, Mount Pearson State Reserve, Doctors Peak Regional Reserve, Cameron Regional Reserve, Mount William National Park, Strzelecki National Park, Patriarchs Conservation Area, Foochow Conservation Area, Wingaroo Nature Reserve, and Killiecrankie Nature Recreation Area. These reserves span the effective geographic range of the species. Jones *et al.* (1999, p. 143) stated that "subsp. *chrysantha* is well represented in reserves, but subsp. *striolata* is represented only in the Strzelecki National Park". This statement is now no longer correct, with the species represented in at least three formal reserves on Flinders Island. Due to imprecise database information, it is likely that the record (labelled "North Patriarch") from the Foochow Conservation Area is actually from the Patriarchs Conservation Area. The record from the Wingaroo Nature Reserve is difficult to place because it is associated with numerous other plant records from the same collector on the same date that extend beyond the boundaries of the reserve, but is most likely from Mount Blyth or Mount Boyes, both low-lying granite rises.

Four records fall within former commercial wood production forests, currently referred to as Future Potential Production Forest (Crown), and only two within areas designated as Permanent Timber Production Zone Land. One of the latter is of very low precision and is more likely from some form of reserved land. The other is from Moorina Hill, first reported in 1970, and confirmed by the author in 1995–2000 as part of a pre-harvest forestry coupe survey.

The species also occurs in one site on private land subject to a conservation covenant under the Tasmanian *Nature Conservation Act 2002*.

There are relatively few records (19 of 109 records in the NVA) on unreserved private freehold, most of which are imprecise and likely refer to nearby reserved land such as the Douglas-Apsley National Park, Strzelecki National Park or reserves in the Bicheno area. One site is on Mount Munro on Cape Barren Island, currently without any formal reservation status. Firth (1965) reported the species from Clarke Island (although no information exists to support this), which is classified as the lungtalanana Indigenous Protected Area.

While reservation *per se* does not impose necessarily ideal management regimes nor preclude potentially deleterious management regimes to a species, in the case of *D. striolata*, the high level of reservation across its range, combined with several separated sites within particular reserves (most notable in reserves such as Freycinet National Park, Douglas-Apsley National Park and Cameron Regional Reserve), appears to offer a reasonably high level of persistence potential and long-term security to the species across its range and sub-regionally.

Conservation status: conservation management

Dockrillia striolata appears to be a generally robust and resilient species, with many "colonies" that are very long-lived. For example, the well-known sites at Bicheno, Freycinet Peninsula and on Flinders Island have been present for over a century. Jones *et al.* (1999, p. 143) suggested that because the "roots spread widely over the rock surfaces and in crevices [they are] not insulated by soil [and] the plants are very vulnerable to all but low-intensity ground fires". However, there is little evidence that the species is deleteriously affected by wildfire or prescribed (fuel reduction) burns. For example, The Hazards on Freycinet Peninsula were subject to an intensive wildfire in February 1980 that left the slopes denuded for many years, exposing many of the massive granite boulders supporting *D. striolata*, but the species remains abundant and healthy across this mountain range.

At a highly localised level, specific "colonies" may be affected by minor land use activities such as the construction and use of walking and mountain bike trails. Again, however, it appears that despite very high visitation that includes visitors scrambling over rocks to gain the best views, "colonies" remain healthy and long-persistent, flowering and fruiting each year, at sites such as Whalers Lookout/Lookout Rock at Bicheno and the lookout on the Wineglass Bay Track on Freycinet Peninsula.

Wood production activities have the potential to include localised sites supporting the species. At a broader level, within the wood production landscape, many sites supporting *D. striolata* would be excluded informally through management of streamside reserves as required by the *Forest Practices Code* (FPA 2015), or through informal reservation through the Management Decision Classification system developed by Forestry Tasmania for publicly managed forests (Orr & Gerrand 1998). At a finer scale of forest management, microhabitats such as rock outcrops are recognised as a site of potential significance for flora in the *Forest Botany Manual* (FPA 2005), the primary planning tool used by forest planners to take account of conservation values in commercial wood production activities. The usual practice is for such habitat features to be excluded from intensive forestry activities. For example, a site on a massive granite outcrop in *Eucalyptus regnans* wet sclerophyll forest at Moorina Hill was detected during a pre-harvest forestry coupe survey (M. Wapstra pers. obs.) and subsequently protected, with a substantial buffer, from surrounding hardwood plantation development.

Conservation status: a review of the formal conservation status of *Dockrillia striolata*

Dockrillia striolata is not presently listed as threatened on either the Commonwealth *Environment Protection and Biodiversity Conservation Act 1999* (EPBCA) or the

Tasmanian *Threatened Species Protection Act 1995* (TSPA). With such a widespread distribution (eastern Tasmania through to northern New South Wales), the species would not qualify for listing under the EPBCA.

However, a review of its potential status on the TSPA is considered warranted because there has been informal concern that former subsp. *chrysantha* may be "possibly threatened" (listing under the NVA), presumably because of its endemic status. Even with the revised taxonomy that no longer recognises the subspecies, the opportunity is taken to assess the taxon against the "*Guidelines for Eligibility for Listing under the* Threatened Species Protection Act 1995" produced by the Scientific Advisory Committee, established under the provisions of the TSPA (DPIW 2008).

Due to its widespread distribution and number of locally abundant subpopulations, there is no notion that the species would qualify as either endangered (Schedule 3) or vulnerable (Schedule 4) under the Act. Following are the criteria for Schedule 5 (rare), copied verbatim from the *Guidelines*, with my comments on their specific application to *D. striolata* in square brackets below each.

A taxon of native flora or fauna may be listed as rare if it has a small population in Tasmania that is not endangered or vulnerable but is at risk (Section 15(4) of the Act).

The following criteria may provide evidence of the level of threat. In order to be considered as rare at least ONE of the criteria A-B should apply.

(A) A taxon of limited distribution or numbers, threatened by existing ongoing processes occurring over sufficient of their range to suggest that they would satisfy the indicative criteria for vulnerable unless the threatening process was abated based on (and specifying) any one of the following:

1. the extent of occurrence is less than 80×80 km or 2000 km^2,

[The extent of occurrence is estimated at 7500 km^2 – SUB-CRITERION NOT MET].

2. the area of occupancy is not more than 0.5 km^2 (50 hectares);

[The area of occupancy is tentatively estimated at no more than a few hectares but the concept of area of occupancy has limited application to this species – SUB-CRITERION REASONABLY NOT MET].

3. taxa that are not A1 or A2 above, but that have very small and localised subpopulations wherever they occur (generally no subpopulation with an area of occupancy greater than 0.01 km^2 (1 hectare) and no more than 1,000 mature individuals).

[While difficult to estimate the abundance of any particular subpopulation, there is no direct evidence of any subpopulation occupying greater than 1 ha and/or more than 1000 mature individuals – SUB-CRITERION MET].

(B) Total population small or restricted and at risk in the form of EITHER of the following:

1. the total population consists of fewer than 10,000 mature individuals, and no more than 2500 mature individuals occur on land that is in an area free from sudden processes capable of causing largely irreversible loss of individuals or habitat; OR

[The total population is estimated in the order of low to high thousands of "colonies", which may technically meet the "fewer than 10,000 mature individuals". However, virtually all subpopulations are considered secure – SUB-CRITERION NOT MET].

2. 90% of mature individuals occur in 15 or fewer subpopulations or locations and no more than 5 of these occur in an area that is free from sudden processes capable of causing largely irreversible loss of individuals or habitat.

[The species is represented by more than 30 subpopulations (or locations), virtually all of which are considered secure – SUB-CRITERION NOT MET].

Evidence points to a species that is widespread, represented by many subpopulations that are well-reserved, resilient to various forms of natural disturbance, and not subject to specific identifiable anthropogenic threats. This statement is applicable to both the Tasmanian mainland part of the population and the Furneaux islands part of the population. However, the species technically meets Criterion A of the *Guidelines* because of the restricted area of occupancy. On this criterion, many Tasmanian vascular and non-vascular plants would qualify for listing. In the author's opinion, a key phrase in the TSPA is "... but is at risk". In terms of predictable threats such as fire, *D. striolata* appears to be a resilient and robust species. It is noted that reservation status *per se* does not form part of these criteria. However, for some species such as *D. striolata*, which occurs in vegetation not requiring specific management intervention to ensure persistence, occurrence in reserves contributes significantly to the concept of "security from risk". This leaves stochastic events as the main threatening process, which by definition are unpredictable. For some species that occur as a single population, stochasticity can come into play. An example is *Azorella macquariensis* (Macquarie Cushion), which occurs on subantarctic Macquarie Island, where it was assumed to be quite secure, before being devastated by a disease, an event that caught everyone by surprise (i.e. genuinely stochastic). In the case of *D. striolata*, the potential impact of a stochastic event is unlikely to manifest as a State-wide whole-of-population crash because of the widespread and geographically separated subpopulations. In the author's opinion, there is insufficient evidence to nominate *D. striolata* as a threatened species under the Tasmanian *Threatened Species Protection Act 1995*.

ACKNOWLEDGEMENTS

Juergen Kellermann (State Herbarium of South Australia) provided information on the collection by A.L. Rogers from the Gordon River, as well as insights into other collections by Rogers. David Ziegeler provided excellent information on his 1970 sighting in the Kelvedon area. Fred Duncan provided information on the erroneous record from the Murdunna Hill area. Judy Rainbird (Queen Victoria Museum) supplied database information of collections at that institute. Miguel de Salas (Tasmanian Herbarium) facilitated access to specimens, provided useful discussions and assisted with the revised synonymy. Perpetua Turner

provided the statistical analyses. Hans and Annie Wapstra, Miguel de Salas and Stephen Harris provided critical reviews of a draft of the manuscript.

REFERENCES

ALA (Atlas of Living Australia) 2019: Occurrence download at https://biocache.ala.org.au/occurrences/search?q=dockrillia%20striolata&fq=state%3A%22Tasmania%22, (accessed on 26 May 2019).

AVH (Australasian Virtual Herbarium) 2019: Occurrence download at https://avh.ala.org.au/occurrences/search?q=lsid%3Ahttp%3A%2F%2Fid.biodiversity.org.au%2Fname%2Fapni%2F166896&qc=data_hub_uid%3Adh9, (accessed on 9 August 2019).

Brieger, F.G. 1981: Subtribe Dendrobiinae. R. Schlechter. *Die Orchideen* **1**(11–12): 636–752.

Buchanan, A.M. 1988: *The Tasmanian Collecting Localities of Ronald Gunn & Joseph Milligan.* Tasmanian Herbarium Occasional Publication No. 1. Tasmanian Herbarium, Hobart: 56 pp.

Clements, M.A. 1989: Catalogue of Australian Orchidaceae. *Australian Orchid Research* **1**: 1–160.

Clements, M.A. & Jones, D.L. 1996: A checklist of the genus *Dockrillia*. *The Orchadian* **12**(2): 60.

Curtis, W.M. 1979: *The Student's Flora of Tasmania Part 4A Angiospermae: Orchidaceae.* Government Printer, Hobart: 138 pp.

de Salas, M.F. & Baker, M.L. 2019: *A Census of the Vascular Plants of Tasmania, including Macquarie Island.* Tasmanian Herbarium, Hobart: 156 pp.

DPIW (Department of Primary Industries & Water) 2008: *Guidelines for Eligibility for Listing under the Threatened Species Protection Act 1995.* Department of Primary Industries & Water, Hobart: 8 pp.

DPIPWE (Department of Primary Industries, Parks, Water & Environment) 2019: *Natural Values Atlas Species Observation download*, (accessed 26 May 2019).

FPA (Forest Practices Authority) 2005: *Forest Botany Manual Module 3 Ben Lomond Region.* Forest Practices Authority, Tasmania: 48 pp.

FPA (Forest Practices Authority) 2015: *Forest Practices Code 2015.* Forest Practices Authority, Tasmania: 120 pp.

Firth, M.J. 1965: *Native Orchids of Tasmania.* C. L. Richmond & Sons Pty. Ltd., Devonport: 90 pp.

Gibberd, J. 1988: Rogers, Richard Sanders (1861–1942). Australian Dictionary of Biography. National Centre of Biography, Australian National University, Canberra. [http://adb.anu.edu.au/biography/rogers-richard-sanders-8259/text14465, published first in hardcopy 1988, (accessed 26 May 2019)].

Harris, S. & Davis, G. 1995: The vegetation and flora of Deal Island, Kent Group. *Papers and Proceedings of the Royal Society of Tasmania* **129**: 43–51.

Harris, S., Buchanan, A. & Connolly, A. 2001: *One Hundred Islands: The Flora of the Outer Furneaux.* Department of Primary Industries, Water & Environment, Hobart: pp. 361.

Hooker, J.D. 1858: *The Botany of the Antarctic Voyage of H.M. Discovery Ships Erebus and Terror. Part III. Flora Tasmaniae Volume II. Monocotyledones and Acotyldones.* Lovell Reeve, London: 378 pp.

Jones, D.L. 1998: Contributions to Tasmanian orchidology. 1: Introduction and the genera *Acianthus, Arthrochilus, Calassodia, Calochilus, Corybas, Cyrtostylis, Dipodium, Dockrillia, Gastrodia, Leptoceras, Microtis, Pyrorchis* and *Townsonia*. *Australian Orchid Research* **3**: 1–15.

Jones, D. 2006: *A Complete Guide to Native Orchids of Australia including the Island Territories.* New Holland Publishers (Australia), Sydney: 496 pp.

Jones, D., Wapstra, H., Tonelli, P. & Harris, S. 1999: *The Orchids of Tasmania.* The Miegunyah Press at Melbourne University Press, Carlton: 317 pp.

Kirkpatrick, J.B. 1995: Vegetation of Dover and Erith islands, Kent Group, Bass Strait. *Papers and Proceedings of the Royal Society of Tasmania* **129**: 25–33.

Mueller, F.M. 1859: *Fragmenta Phytographiae Australiae Vol. I.* Auctoritate Gubern. Coloniæ Victoriæ, Ex Officina Joannis Ferres, Melbourne: 252 pp.

NCHD (Natural and Cultural Heritage Division) 2014: *lungtalanana (Clarke Island), Natural Values Survey 2014.* Nature Conservation Report Series 15/2, Hamish Saunders Memorial Trust, New Zealand & Natural and Cultural Heritage Division, Department of Primary Industries, Parks, Water & Environment, Hobart: 158 pp.

Orr, S. & Gerrand, A.M. 1998: Management Decision Classification: a system for zoning land managed by Forestry Tasmania. *Tasforests* 10: 1–14.

Rodway, L. 1903: *The Tasmanian Flora.* Government Printer, Hobart: 320 pp.

Spicer, W.W. 1878: *A Handbook of the Plants of Tasmania.* J. Walch and Sons, Hobart Town: 160 pp.

Wapstra, H., Wapstra, A., Wapstra, M. & Gilfedder, L. 2005: *The Little Book of Common Names for Tasmanian Plants.* Department of Primary Industries, Water & Environment, Tasmania: 88 pp. [updated online annually]

WCSP 2019: *World Checklist of Selected Plant Families.* Facilitated by the Royal Botanic Gardens, Kew. [http://wcsp.science.kew.org, (accessed 26 May 2019)].

(accepted 30 October 2019)

APPENDIX 1 — Specimens examined[1]

AUSTRALIA: Tasmania: Mt William, W face, Mt William National Park, 27 Oct 1981, *M.J. Brown 6* (HO55660), [7]; near Georges Bay, Jan 1896, *L. Rodway s.n.* (HO500736), [4]; Bicheno, Sep 1927, *F. Perrin s.n.* (HO503341), [5]; Bicheno, Lookout Rock, Jan 1984, *P.A. Collier 22* (HO115674), [3]; Mt Strzelecki, lower quarter of track, Flinders Island, 12 Jan 2007, *M. Visoiu 265, L. Perrins & A.M. Buchanan* (HO543662) [0, no flowers present on collection]; North Patriarch, 30 km N of Lady Barron, Flinders Island, 26 Oct 1990, *P.A. Collier 4872* (HO127074), [5], Mt Peter, summit, 29 Dec 1985, *A.M. Buchanan 7686* (HO406932), [0, no flowers present on collection]; Cube Rock, SE slopes of Mt Cameron, *A. Moscal 4088* (HO101481), [10]; Mt Strzelecki track, Flinders Island, *P.A. Collier 780* (HO95849), [6]; Mt Killiecrankie, nearly opposite Sisters Isles, 4 Oct 1847, *J. Milligan 923* (HO500737), [2]; Mt Munro, Cape Barren Island, 7 Oct 1988, *P.A. Collier 3574* (HO118398), [5]; Apsley River, 2 Nov 1985, *P.A. Collier 948* (HO116804), [7]; Mt Cameron, 18 Nov 1983, *A. Moscal 4180* (HO94553), [17]; Mt Cameron, 21 Oct 1983, *A. Moscal 3693* (HO82040), [9]; Mt William, summit, 8 Sep 1983, *A. Moscal 2563* (HO70862), [3]; Bayleys Hill, summit ridge, 16 Sep 1983, *A. Moscal 2758* (HO71758), [5]; Georges Bay, Nov 1890, *L. Rodway 750* (HO500769), [6]; Second Sugarloaf, 2.5 km south east of Gladstone, 29 Sep 1988, *F. Coates s.n.* (HO112450), [4]; Bicheno, Harveys Farm Road, 23 Oct 1992, *H. Wapstra & A. Wapstra s.n.* (HO329686), [2]; Bedggood Hill, near Bicheno, 20 Mar 1980, *A. Moscal 234* (HO34275), [0, no flowers present on collection]; Wineglass Bay, Oct 1940, *W.M. Curtis s.n.* (HO500767),[1]; Strzelecki Track, Flinders Island, *H. Wapstra & A. Wapstra s.n.* (HO518504), [8]; Bicheno, NE of town centre, *D.L. Jones 13582 & B.E. Jones* (HO523462, TYPE of subsp. *chrysantha*), [3]; Bicheno, 5 Nov 1962, *no collector indicated* (HO537072), [4]; Wineglass Bay Track, Freycinet National Park, 25 Oct 2008, *J.K. Janes 405* (HO549725), [2]; Coles Bay, Sep 1948, *W.M. Curtis s.n.* (HO536589), [6]; Coles Bay, Oct 1951, *G. Sharman s.n.* (HO536590), [5]; Lanka Road area, near the Great Musselroe River, 5 Oct 2005, *M. Douglas s.n.* (HO533819), [2]; NE end of Mt Cameron Range, 27 Nov 2005, *M. Douglas s.n.* (HO535874), [3]; Bicheno, 23 Oct 1969, *C. Shea s.n.* (HO563452), [7]; Flinders Island, Mt Strzelecki, 19 Oct 1976, *M. Allan s.n.* (HO569976), [4]; Freycinet National Park, Wineglass Bay Track, 25 Oct 1980, *M. Allan s.n.* (HO589553), [6]; Mt Cameron East, 22 Oct 1973, *C. Taylor s.n.* (HO589554), [2]; Coles Bay, Oct 1957, *H. Gulline s.n.* (HO500768), [1]; East Coast, Oct 1931, *J. Overall 7* (HO105721), [1].

[1] Location (as per HO collection label), collector, date of collection, HO accession no., [number of flowers measured from collection sheet]

PATTERNS AND CAUSES OF MARSUPIAL PATHS IN SUBALPINE TASMANIA

by Emma C. Nichols and Jamie B. Kirkpatrick

(with seven text-figures, two plates and one table)

Nichols, E.C. & Kirkpatrick, J.B. 2019 (14:xii) Patterns and causes of marsupial paths in subalpine Tasmania. *Papers and Proceedings of the Royal Society of Tasmania* 153: 53–60. ISSN 0080-4703. Geography and Spatial Sciences, University of Tasmania, Private Bag 78, GPO, Hobart, Tasmania 7001, Australia (ECN, JBK*). *Author for correspondence. Email: j.kirkpatrick@utas.edu.au

Animal paths can improve connectivity of species, alter vegetation growth, reduce fuel loads and further our knowledge of the behavioural ecology of mammals. Path patterning and the motivations for path creation are not well understood. The present study tested the hypotheses that animals form paths: (i) to efficiently move between palatable patches; (ii) for access to water; (iii) for access to shelter and (iv) to avoid predators. We used high-definition aerial online imagery to map paths in five treeless subalpine environments in Tasmania. Surveys of vegetation and scat counts were conducted at each site. We found that locality influenced wallaby scat density and that macropods and wombats created direct paths to move between areas of palatable vegetation, shelter and water. There was some weak indication of predator avoidance in some of the patterning. However, shelter from cold, strong southwesterly winds is a feasible alternative motivation for the patterns.

Key Words: landscape of fear, macropod, path, shelter, wombat.

INTRODUCTION

Animal paths occur throughout many terrestrial environments and are often a prominent feature of natural landscapes. Animal paths can help identify patterns and processes of movement that can suggest the underlying motivations for path usage (Kays *et al.* 2015). Understanding path usage can therefore improve our knowledge of the behavioural ecology of a species and of the effects of animal paths on other organisms. Animal paths can influence plant growth (Ferguson *et al.* 2010), improve connectivity and limit fire spread (Ferguson *et al.* 2010; Knapp *et al.* 2016).

Studies of the movements of terrestrial mammals that create paths have predominantly been of large hooved mammals, such as cattle (Ganskopp *et al.* 2000; Depew 2005), White-tailed Deer (*Odocoileus virginianus*; Depew 2005), Asiatic Wild Ass (*Equus hemionus*; Davidson *et al.* 2013), Wildebeest (*Connochaetes taurinus*), Tibetan Wild Ass (*Equus kiang*; Sharma *et al.* 2004) and Woodland Caribou (*Rangifer tarandus caribou*; Johnson *et al.* 2002). Non-hard-hooved large herding or pack-forming mammals such as the African Savannah elephants (*Loxodonta africana*; Loarie *et al.* 2009) and Grey Wolf (*Canis lupus*; Huck *et al.* 2011) also form paths. Davidson *et al.* (2013) and Sharma *et al.* (2004) found that wild asses avoid steep slopes. Vegetation density played a significant role in movement decision-making in cattle and White-tailed Deer (Depew 2005), as well as the Asiatic Wild Ass (Davidson *et al.* 2013). Conserving energy by following paths of least effort is also suggested to be a major influence on cattle movements, particularly when in search of water (Ganskopp *et al.* 2000). Some of the influences on wildebeest movements are thought to be water access, level of predation risk and sufficient access to food (Martin *et al.* 2015). The only study that focused on the effects of soft-footed mammal movements found that repetitive movements by wombats, wallabies, pademelons and rats reduced competition for plant species that would otherwise be outcompeted and that paths occupied up to 12% of the ground within particular vegetation types (Ferguson *et al.* 2010).

As well as path creation to conserve energy, mammals may create concealed paths to avoid predators in a landscape of fear (Laundré *et al.* 2001). The landscape of fear results from the non-lethal effects of the fear of predation by prey species. The landscape of fear has been observed in behavioural changes in both elk (*Cervus elaphus*) and bison (*Bison bison*) following the reintroduction of wolves (*Canis lupus*) to Yellowstone National Park (Laundré *et al.* 2001). This behavioural change showed that prey species adjusted their time spent foraging in a location, as well as avoiding locations associated with higher predation risk (Laundré *et al.* 2001).

Most of the limited work on animal paths has not mapped the paths themselves. Where paths have been mapped (Ganskopp *et al.* 2000; Davidson *et al.* 2013), they have been related to topography rather than vegetation type.

The aim of the present study was to determine patterns and processes in path formation by Australian marsupials in a variety of high-altitude, subalpine environments. We tested the hypothesis that animals form paths to efficiently move between palatable patches, water and shelter. We also determined if there is any evidence that paths form on routes that allow animals to avoid predators.

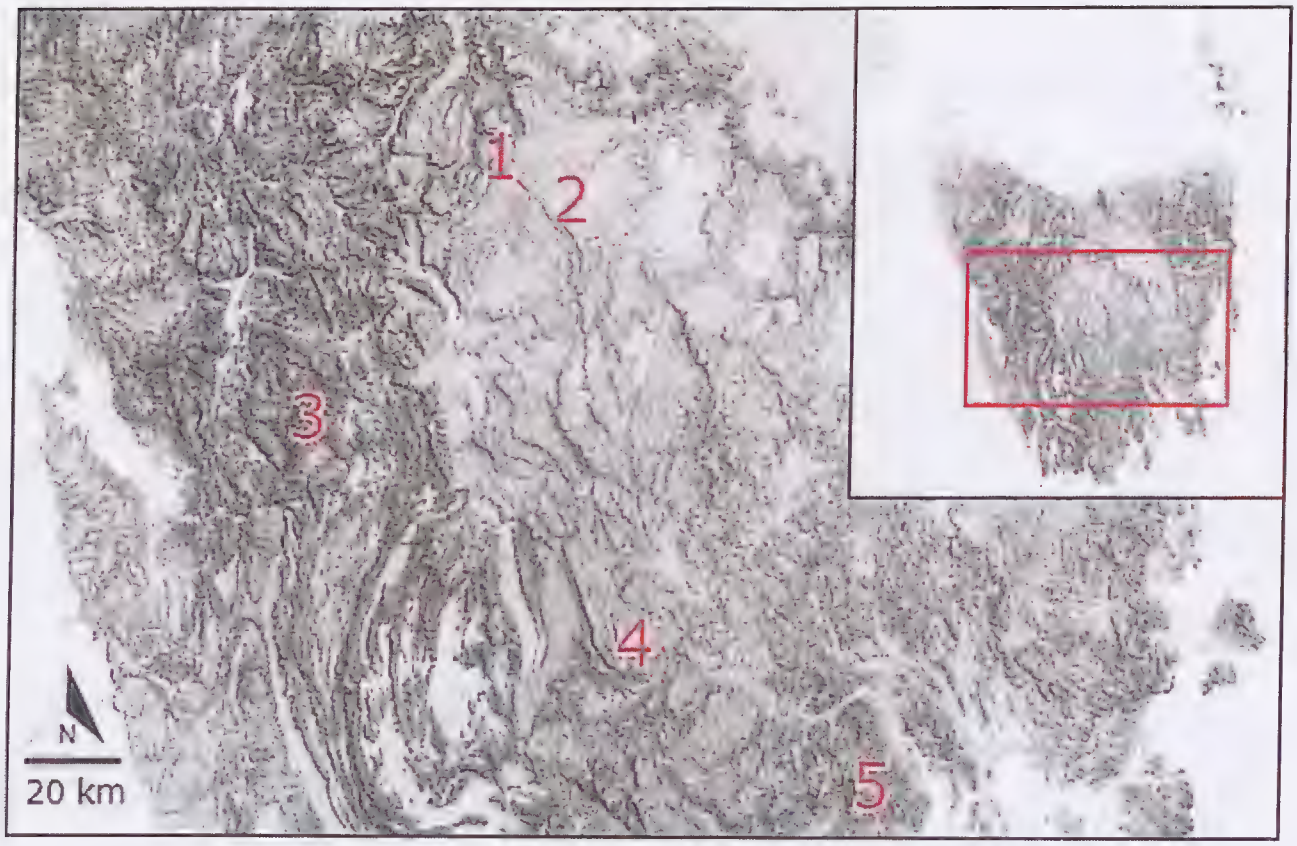


FIG. 1 — Locations of mapping. 1 – Walls of Jerusalem National Park. 2 – Lake Ada, Central Plateau. 3 – Frenchmans Cap. 4 – Mount Field East. 5 – Mount Wellington Plateau. Source: The LIST Map (2019).

METHODS

Study areas

Five subalpine locations were chosen for surveying because of the availability of high-definition online aerial imagery, which clearly showed paths and vegetation types at each site (fig. 1). The Land Information System Tasmania Map (The LIST Map 2019) was used to identify potential study areas, which were selected to cover a range of path patterns and vegetation types.

The Mount Wellington site (GDA94, MGA55, E517376, N5250586) is on a subalpine plateau at 1140 m. The site rests upon Jurassic dolerite (The LIST Map 2019), and the average annual rainfall is 1000–1500 mm (Bureau of Meteorology 2016).

Two sites were chosen within the Walls of Jerusalem (E442128, N5370172; E441326, N5370406). The first site was on a north-facing slope where the elevation peaked at 1280 m. The second site was on flat ground, at the mouth of Lake Solomon, at 1260 m. The sites are on Jurassic dolerite (The LIST Map 2019). Annual rainfall averages 2000–3000 mm (BOM 2016).

Lake Ada in the Central Plateau Conservation Area (E455621, N5362641) is at 1140 m. Glacio-fluvial and aeolian material derived from dolerite form rich soils (Pharo & Kirkpatrick 1994). Annual rainfall averages 1000–1500 mm (BOM 2016).

A site was selected below Mount Field East, in the Mount Field National Park (E471116, N5276984) where the elevation reaches 1190 m along a flat surface. This site is underlain by Jurassic dolerite (The LIST Map 2019), with average annual rainfall of 2000–3000 mm (BOM 2016).

Frenchmans Cap is an alpine region with consistently cool temperatures all year round. This site is within the Franklin Gordon Rivers National Park (E404295, N5320029). The elevation reaches 1040 m. The surface geology is quartzite (The LIST Map 2019), and the site has an annual average rainfall of 2000–3000 mm (BOM 2016).

Data collection

Maps of paths and vegetation types were produced at each location from high-resolution aerial photographs from the Land Information System Tasmania Map (The LIST Map 2019) and ground truthing. The mapping scale for Frenchmans Cap and the Walls of Jerusalem was 1:846. All other locations were mapped at 1:1693.

The following vegetation types were discriminated: *Astelia alpina* sedgeland, *Poa* grassland, bog, rushland, cushion heath, *Orites* heath, *Gleichenia* fernland, *Sphagnum* bog, *Richea acerosa* heath, *Richea scoparia* heath, *Empodisma minus* rushland, *Deschampsia cespitosa* tussock grassland, *Orites acicularis* tall shrubland, *Athrotaxis cupressoides* (Pencil Pine) scrub, *Athrotaxis* scrub, and *Eucalyptus coccifera* scrub.

Scat surveys were conducted at each location. Typical areas for each of the main vegetation types were selected. Scats were counted in ten 1×1 m quadrats randomly located within a 10×10 m sample area. Scats were counted by taxon: macropods (Bennets Wallaby (*Macropus rufogriseus*) and Tasmanian Pademelon (*Thylogale billardierii*)), Bare-nosed Wombat (*Vombatus ursinus*), Brush-tailed Possum (*Trichosurus vulpecula*), Spotted-tailed Quoll (*Dasyurus maculatus*), European Rabbit (*Oryctolagus cuniculus*) and native rodents (*Rattus* spp.).

Statistical analysis

Wallaby scat counts were used for statistical analyses, as wallaby scats were abundant across each site (other taxa were present at only one or two sites). One-way ANOVA was used to assess whether locality had an effect on wallaby scat density, as well as whether there was an effect of vegetation type on wallaby scat density. Each vegetation type was compared to all the others.

The map of the area mapped on Mount Wellington was used to test whether paths were randomly located. The vegetation type at each end of each path was recorded. An equal number of points was distributed randomly across the map and the vegetation type at each of these points was recorded. The significance of the deviation of the path end points from a random distribution was tested using Fishers exact probability test. All analyses used Minitab (version 18.1).

RESULTS

Wallaby scat density averaged 12.4 scats per m^2. Wallaby scat density was lower at Frenchmans Cap (1.5 per m^2) than in the other areas (10.4–18.8 per m^2, ANOVA $F = 11.77_{6,208}$, $P < 0.001$). Wallaby scats were also denser in vegetation with *Poa* than in vegetation without *Poa* as a dominant (6.8 per m^2 cf. 0.2 per m^2, ANOVA $F = 13.71_{1,208}$, $P < 0.001$). No other vegetation type had higher or lower scat densities than any other vegetation type.

The length of paths through Mount Wellington rushland was 1532 m (fig. 2). There were 144 m of paths in vegetation dominated by *Poa* spp. and *Astelia alpina*, and 36 m of paths in grazed rushland. The ends of paths at Mount Wellington were associated with vegetation with *Poa* spp. ($p = 0.023$) and avoided *Richea scoparia* heath and scrubland ($p = 0.005$) (table 1). There was some indication that the paths accessed shelter and water, as well as more palatable vegetation, which included an inexplicably heavily grazed patch of *Empodisma minus* (pl. 1), a species largely ungrazed elsewhere on Mount Wellington.

At the Walls of Jerusalem (figs 3, 4) and Lake Ada (fig. 5), paths connected patches of palatable vegetation and these patches with shelter in scrub or woodland, largely passing through alpine heath. In both places, *Astelia alpina* was grazed when mixed with, or adjacent to, grassland (pl. 2), which was not the case on Mount Wellington (fig. 2), Mt Field East (fig. 6) or Frenchmans Cap (fig. 7). At the Walls of Jerusalem and Lake Ada, there were paths that ran through concealing vegetation parallel to the most palatable vegetation types (figs 3–5). These tracks are located to the northeast of the palatable vegetation.

At Mt Field East, the *Orites* heath and the *Poa* grassland were the main areas of palatable plants. Paths were evident within the heath as well as connecting the heath to sheltering vegetation (fig. 6). Only one small length of path associated with a rock outcrop (fig. 7) was mapped on Frenchmans Cap.

DISCUSSION

The lower level of scat counts at Frenchmans Cap than elsewhere is likely related to low soil fertility resulting from its quartzite substrate and high rainfall (Kirkpatrick *et al.* 2014). Highly palatable vegetation with *Poa* is absent. As with bushwalkers' paths (Whinam & Chilcott 2003), there appears to be a threshold in numbers before paths become apparent. The importance of grassland in supporting macropods in these subalpine areas is consistent with State-wide data (Kirkpatrick *et al.* 2011, 2016, Styger *et al.* 2011).

Macropods and wombats are largely responsible for path formation. They comprise most of the vertebrate animal biomass, as indicated by scat counts (Ferguson *et al.* 2010). Paths tend to be highly visible within heath and rushland, perhaps indicating that the animals spread out in the more open palatable vegetation types and that any paths are hidden from view in the scrub and woodland where macropods shelter during the day. The paths appear largely to be least-effort routes, similar to the wild ass paths documented by Davidson *et al.* (2013).

The paths connect grazed areas and also grazed areas with shelter. However, on Mount Wellington, one path ends at permanent water, and some of the paths at Lake Ada and the Walls of Jerusalem could be interpreted as a product of a landscape of fear in that they parallel palatable vegetation but lie within concealing bushes. These two sites have the largest extents of palatable vegetation types and the highest concentrations of scats, and so support many carnivores such as the Tasmanian Devil (*Sarcophilus harrisii*) and Wedge-tailed Eagle (*Aquila audax*). Shooting of wallabies roused by dog packs is occasionally allowed near Lake Ada (Department of Primary Industries, Parks, Water and Environment 2016). However, we cannot conclude with certainty that the tracks parallel to the grazed areas are a product of fear (c.f. Laundré *et al.* 2001), as the grasslands are occasionally immersed and are more exposed to chilling winds than paths among shrub shelter. The location of the parallel tracks to the northeast of palatable vegetation strongly suggests shelter from winds as a motive, given the known importance of strong, icy southwesterly winds in moulding the subalpine vegetation (Morgan *et al.* 2010).

This present study appears to be the first that maps animal path patterning by Tasmanian herbivorous marsupials. We conclude that animals form paths to efficiently move between palatable patches, water and shelter, but suggest that shelter from the elements rather than from predators may be responsible for paths parallel to palatable vegetation.

TABLE 1 — Counts of animal paths' beginning and end points at Mount Wellington

	Poa grassland	*Eucalyptus coccifera* scrub	Rushland	String Bog	Water	Grazed rushland	*Richea scoparia* heath/scrub
Path	*9*	0	15	0	0	3	0
Non-path[1]	*3*	1	14	2	0	0	7

[1] Non-path shows the random count.

PLATE 1 – Heavily grazed patch of *Empodisma minus* at Mount Wellington, with ungrazed *Astelia alpina*.

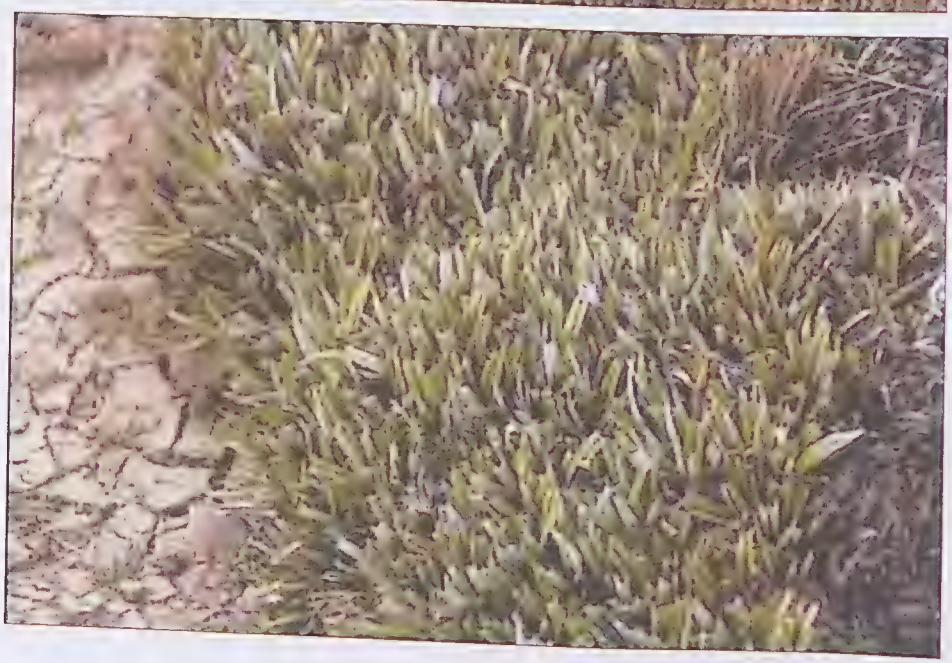


PLATE 2 — Grazed *Astelia alpina* at Lake Ada.

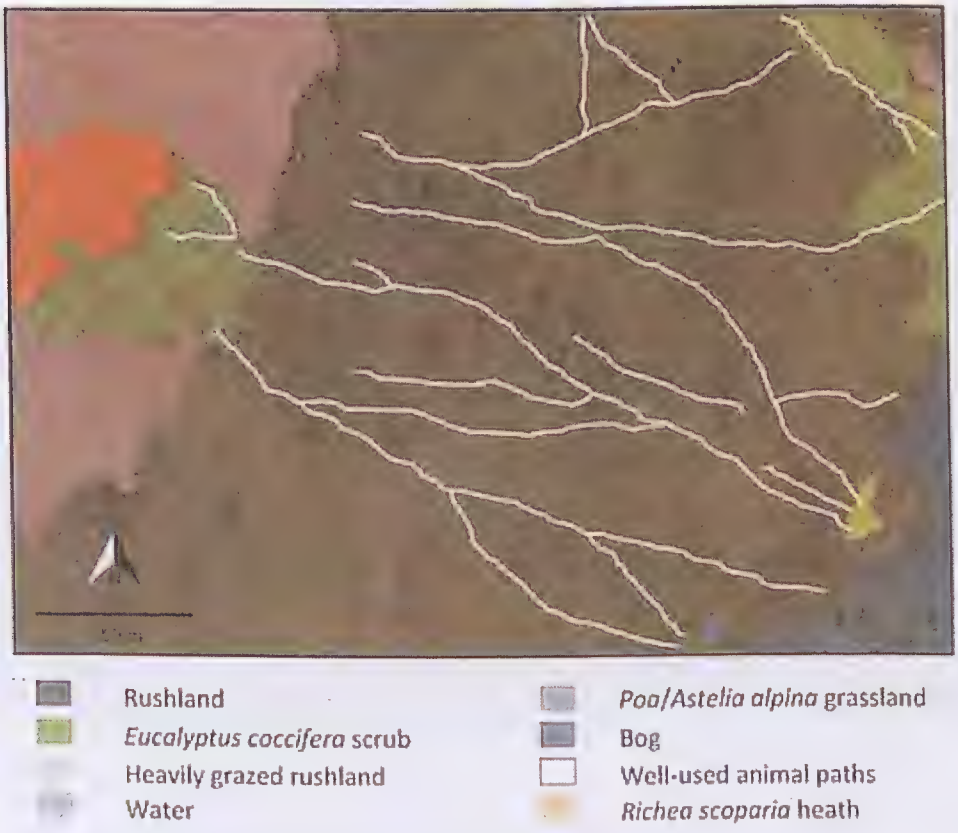


FIG. 2 — Mount Wellington Plateau (E517482, N5250552). Marsupial paths running through rushland, leading to water sources and palatable vegetation. Source: The LIST Map (2019).

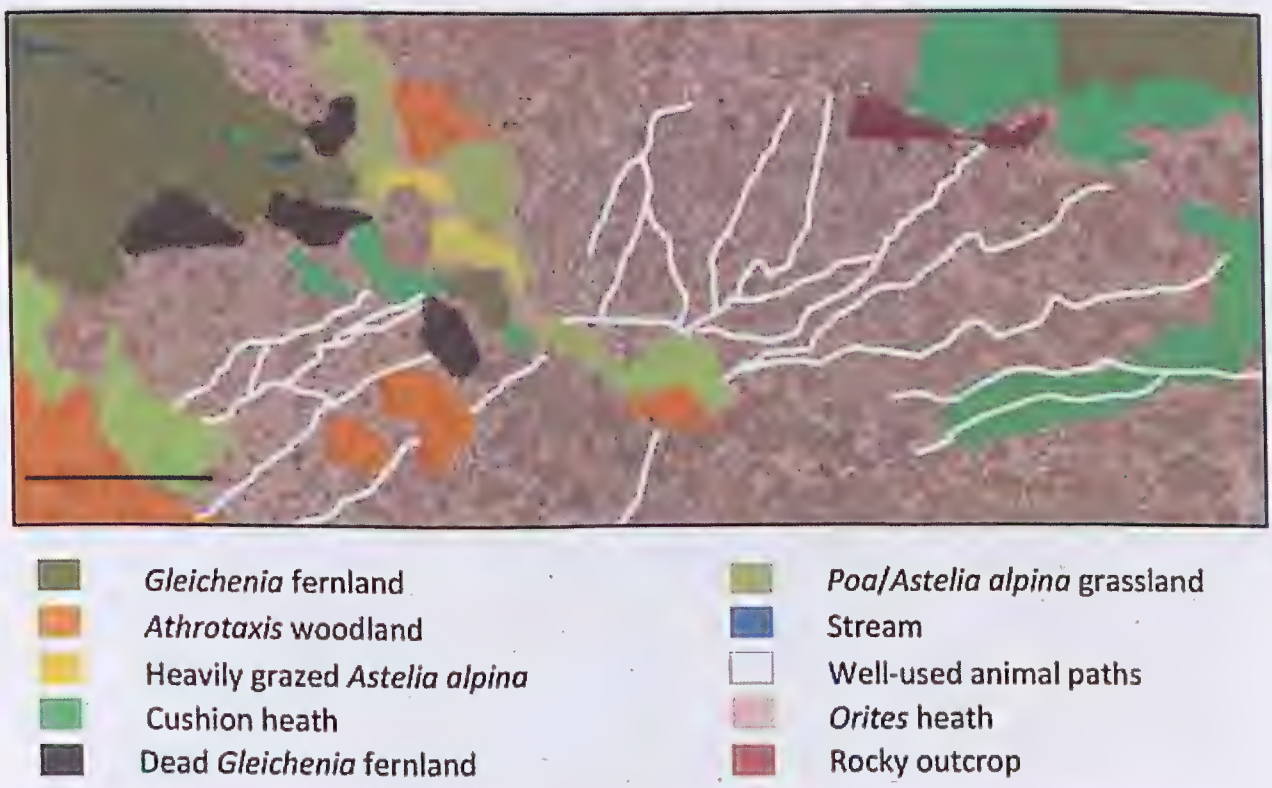


FIG. 3 —Walls of Jerusalem (E442039, N5370177). Dense paths running though *Orites* heath. Source: The LIST Map (2019).

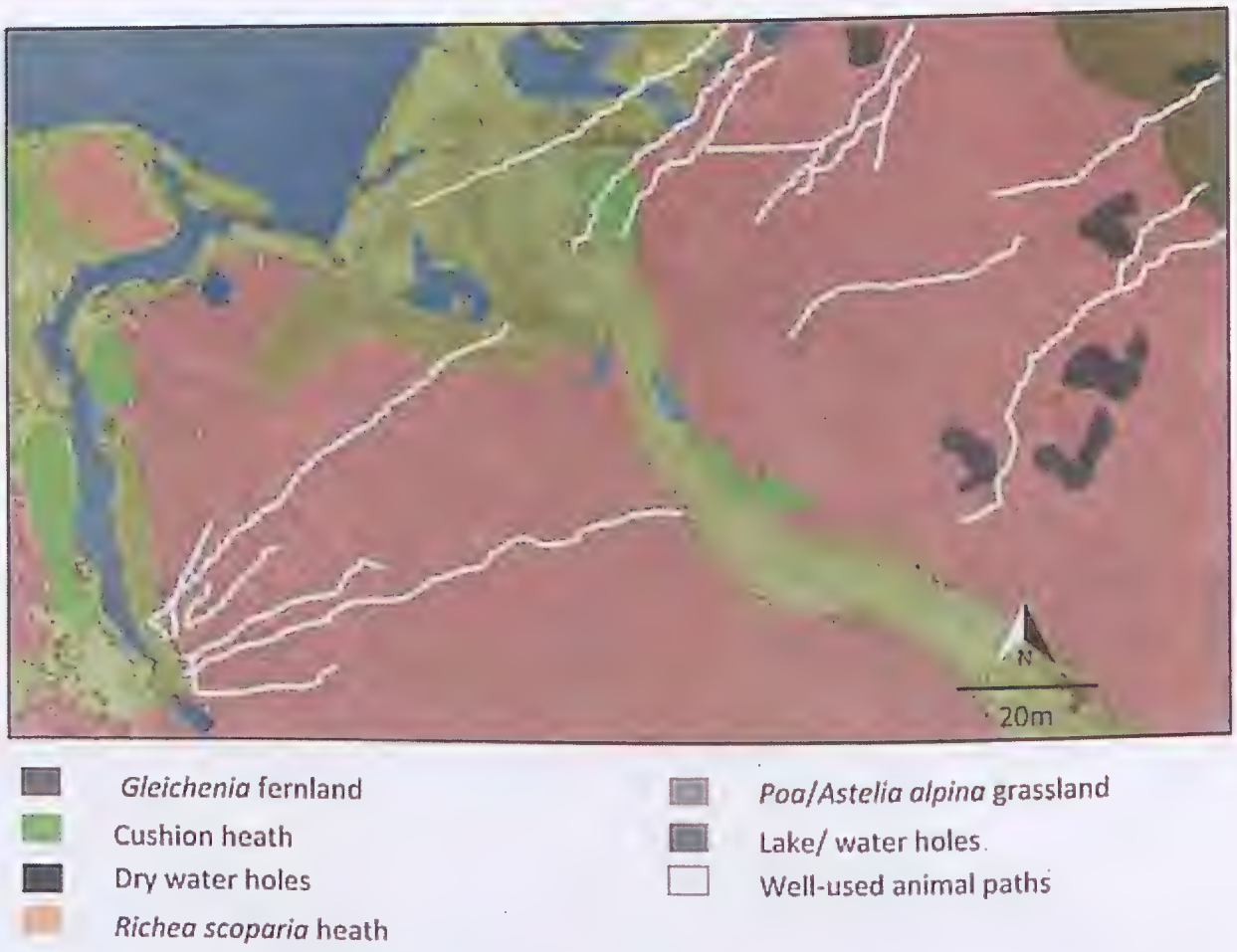


FIG. 4 —At the mouth of Lake Solomon, Walls of Jerusalem National Park (E441326, N5370406). Paths running predominantly west to east. Source: The LIST Map (2019).

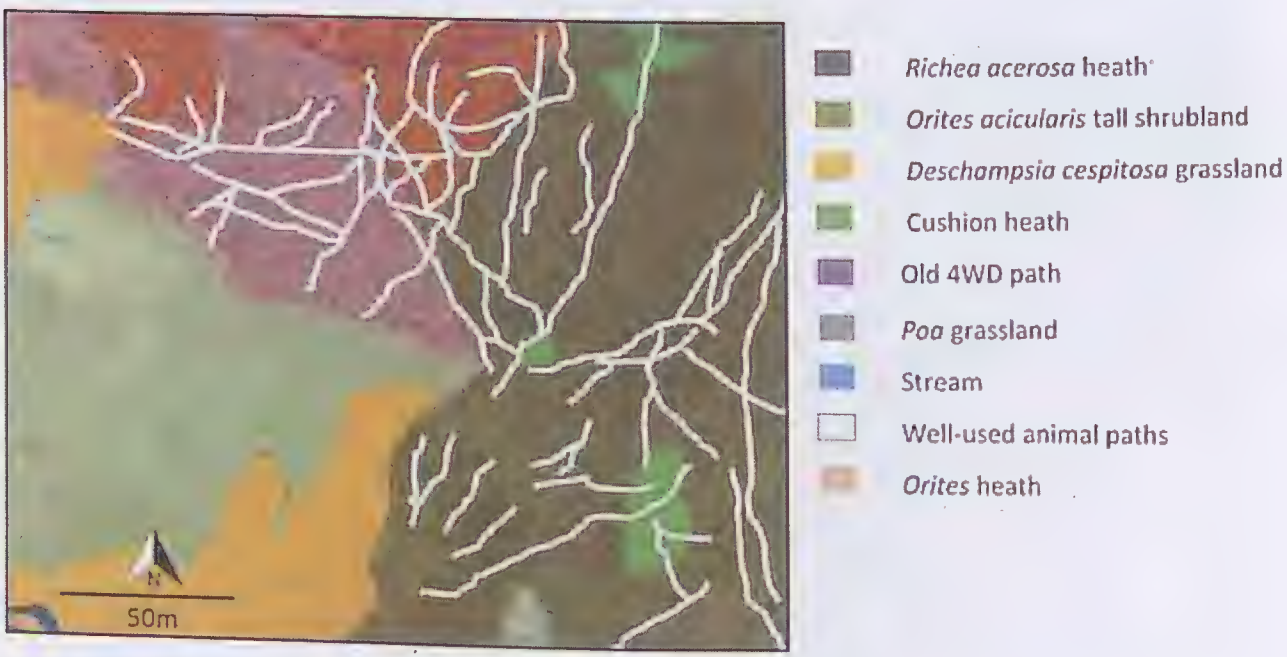


FIG. 5 — Lake Ada, central Highlands (E455621, N5362641). Paths running through a variety of dense vegetation types. Source: The LIST Map (2019).

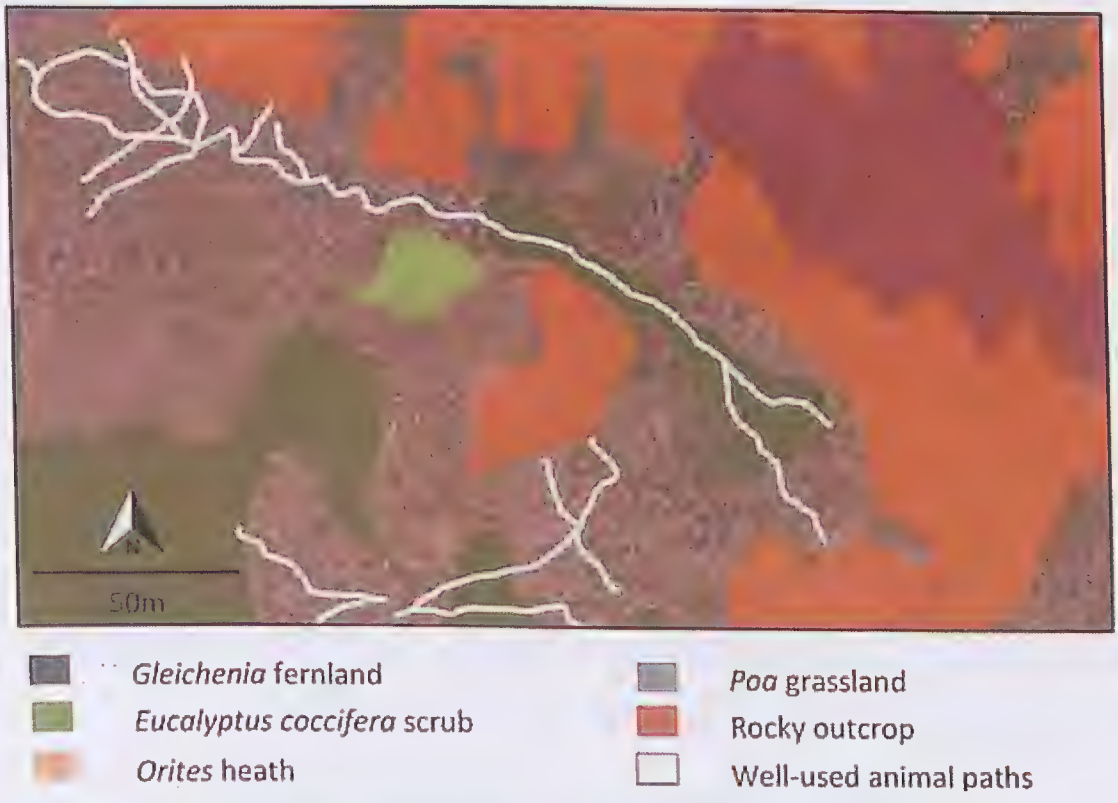


FIG. 6 —Mount Field East (E470468, N5277210). Paths running predominantly east to west at the scrub edge. Source: The LIST Map (2019).

FIG. 7— Frenchmans Cap (E404295, N5320029). One visible path travelling down north-facing slope. Source: The LIST Map (2019).

REFERENCES

BOM (Bureau of Meteorology) 2016: Bureau of Meteorology average annual, seasonal and monthly rainfall. http://www.bom.gov.au/jsp/ncc/climate_averages/rainfall/index.jsp?period=an&area=ta#maps (accessed 23 May 2019).

Davidson, A., Carmel, Y. & Bar-David, S. 2013: Characterizing wild ass pathways using a non-invasive approach: applying least-cost path modelling to guide field surveys and a model selection analysis. *Landscape Ecology* **28**: 1465–1478.

Department of Primary Industries, Parks, Water and Environment 2016: Tasmanian Wilderness World Heritage Management Plan 2016. https://dpipwe.tas.gov.au/ conservation/tasmanian-wilderness-world-heritage-area-(twwha)/twwha-management-plan (accessed 30 May 2019).

Depew, J.J. 2005: *Habitat selection and movement patterns of cattle and white-tailed deer in a temperate savanna.* Doctoral dissertation, Texas A&M University.

Ferguson, A., Kirkpatrick, J. & Pharo, E. 2010: Variability in the incidence and characteristics of animal paths in montane vegetation. *Plant Ecology & Diversity* **3**: 9–17.

Ganskopp, D., Cruz, R. & Johnson, D.E. 2000: Least-effort pathways? A GIS analysis of livestock trails in rugged terrain. *Applied Animal Behaviour Science* **68**: 179–190.

Huck, M., Jędrzejewski, W., Borowik, T., Jędrzejewska, B., Nowak, S. & Mysłajek, R.W. 2011: Analyses of least cost paths for determining effects of habitat types on landscape permeability: wolves in Poland. *Acta Theriologica* **56**: 91–101.

Johnson, C.J., Parker, K.L., Heard, D.C. & Gillingham, M.P. 2002: A multiscale behavioral approach to understanding the movements of woodland caribou. *Ecological Applications* **12**: 1840–1860.

Kays, R., Crofoot, M.C., Jetz, W. & Wikelski, M. 2015: Terrestrial animal pathing as an eye on life and planet. *Science* **348**: DOI: 10.1126/science.aaa2478.

Kirkpatrick, J.B., Marsden-Smedley, J.B. & Leonard, S.W.J. 2011: Influence of grazing and vegetation type on post-fire flammability. *Journal of Applied Ecology* **48**: 642–649.

Kirkpatrick, J.B., Green, K., Bridle, K.L. & Venn, S.E. 2014: Patterns of variation in Australian alpine soils and their relationships to parent material, vegetation formation, climate and topography. *Catena* **121**: 186–194.

Kirkpatrick, J.B., Marsden-Smedley, J.B., di Folco, M-B. & Leonard, S.W.J. 2016: Influence of grazing and vegetation type on post-fire floristic and lifeform composition in Tasmania. *Plant Ecology* **217**: 57–69.

Knapp, S., Stadler, J., Harpke, A. & Klotz, S. 2016: Dispersal traits as indicators of vegetation dynamics in long-term old-field succession. *Ecological Indicators* **65**: 44–54.

Laundré, J.W., Hernández, L. & Altendorf, K.B. 2001: Wolves, elk, and bison: reestablishing the "landscape of fear" in Yellowstone National Park, USA. *Canadian Journal of Zoology* **79**: 1401–1409.

Loarie, S.R., Van Aarde, R.J. & Pimm, S.L. 2009: Fences and artificial water affect African savannah elephant movement patterns. *Biological Conservation* **142**: 3086–3098.

Martin, J., Benhamou, S., Yoganand, K. & Owen-Smith, N. 2015: Coping with spatial heterogeneity and temporal variability in resources and risks: adaptive movement behaviour by a large grazing herbivore. *PloS ONE* **10**: e0118461.

Morgan, S., Kirkpatrick, J.B. & di Folco, M.-B. 2010: Wind-controlled linear patterning and cyclic succession in Tasmanian *Sphagnum* mires. *Journal of Ecology* **98**: 583–591.

Pharo, E.J. & Kirkpatrick, J.B. 1994: Vegetation of the alpine sand dunes at Lake Augusta, Tasmania. *Australian Journal of Ecology*, **19**: 319–327.

Sharma, B.D., Clevers, J., De Graaf, R. & Chapagain, N.R. 2004: Mapping *Equus kiang* (Tibetan Wild Ass) habitat in Surkhang, Upper Mustang, Nepal. *Mountain Research and Development* **24**: 149–157.

Styger, J.K., Kirkpatrick, J.B., Marsden-Smedley, J.O.N. & Leonard, S.W.J. 2011: Fire incidence, but not fire size, affects macropod densities. *Austral Ecology* **36**: 679–686.

The LIST Map 2019: The Land Information System Tasmania map, Department of Primary Industries, Parks, Water and Environment (version 1.2.3-21). https://maps.thelist.tas.gov.au/listmap/app/list/map (accessed 25 May 2019).

Whinam, J. & Chilcott, N.M. 2003: Impacts after four years of experimental trampling on alpine/sub-alpine environments in western Tasmania. *Journal of Environmental Management* **67**: 339–351.

(accepted 6 November 2019)

VEGETATION COMMUNITIES AND EDAPHIC RELATIONSHIPS ALONG A TYPICAL COASTAL SALTMARSH TO WOODLAND GRADIENT IN EASTERN TASMANIA

by John Aalders, Peter McQuillan and Vishnu Prahalad

(with five text-figures and six tables)

Aalders, J., McQuillan, P. & Prahalad, V. 2019 (14:xii). Vegetation communities and edaphic relationships along a typical coastal saltmarsh to woodland gradient in eastern Tasmania. *Papers and Proceedings of the Royal Society of Tasmania* 153: 61–74. ISSN 0080-4703.
Geography and Spatial Sciences, School of Technology, Environments and Design, University of Tasmania, Private Bag 78, GPO, Hobart, Tasmania 7001, Australia (JA*, PM, VP). *Author for correspondence. Email: John.Aalders@utas.edu.au

Saltmarsh soils impose harsh selection pressures on vegetation resulting in characteristic plant communities. For our study of the effect of edaphic factors on vegetation we chose Long Point in Moulting Lagoon, Tasmania's largest saltmarsh, which is dominated by a diverse assemblage of halophytic succulents and graminoids. Three transects were established to sample variations in vegetation along the gradient from saltmarsh to woodland. Soil samples were analysed for summer and winter moisture, pH, and electrical conductivity (EC); a mixed summer and winter sample from each point was analysed for soil organic matter (SOM) and carbon. Additionally, a particle size analysis was carried out on all summer samples. Aspects of soil characteristics were aligned to classified vegetation groups and elevation. Moisture, pH, EC, SOM and carbon were all negatively correlated with elevation; the saltmarsh zone displaying higher levels of all variables than those in the adjacent woodland zone. Clay content decreased and sand content increased from the marine margin of the saltmarsh zone to the woodland zone. Within the saltmarsh zone, soil moisture, EC and carbon had highest values in the low marsh area, with values decreasing towards the upper marsh area. This study deepens our understanding of the roles various edaphic factors play in the floristic composition of coastal saltmarshes.

Key Words: Edaphic factors, elevation, soil carbon, soil-vegetation relations, zonation, coastal saltmarsh.

INTRODUCTION

Saltmarshes are defined by plant species that are adapted to a harsh environment characterised by high salinity, regular to intermittent inundation, as well as exposure to fluctuating temperatures, winds and waves (Adam 1990, Bridgewater & Cresswell 2003, Saintilan 2009a, Prahalad *et al.* 2015). Within this distinctive ecological habitat, the spatial distribution of plants is organised into characteristic communities arranged in a sequence or zonation determined by fine-scale changes in a broad array of local environmental variables (Long & Mason 1983, Silvestri *et al.* 2005). Many authors have described saltmarsh vegetation communities at different spatial scales, from continental (in Australia: Bridgewater & Cresswell 1999, 2003, Saintilan 2009a) to local (in Tasmania: Kirkpatrick & Glasby 1981, Prahalad *et al.* 2011; in Victoria: Boon *et al.* 2015).

At a local ecological scale, attention has been largely focussed on the distribution and patterns of variation in vegetation (Adam 2002), often with limited reference to the range of edaphic factors including salinity, moisture, pH, soil organic matter (SOM) and carbon (Snow & Vince 1984, Vince & Snow 1984, Álvarez-Rogel *et al.* 2000, Huckle *et al.* 2000, Silvestri *et al.* 2005, Anisfeld 2012). During the past two decades, with an increasing focus on conservation and restoration, a renewed global interest in saltmarsh soils has emerged (Anisfeld 2012, Burden *et al.* 2013, Kelleway *et al.* 2016). In the Australian context, except for the pioneering work by Clarke & Hannon (1967, 1969, 1970), the focus of most recent studies has been on sedimentary dynamics (Rogers *et al.* 2013) or carbon content of saltmarsh soils (Kelleway *et al.* 2016). Little attention has been given to the range of other edaphic factors generally considered in soil-vegetation relations (Huckle *et al.* 2000, Landi & Angiolini 2015). As a consequence, the determinants of fine-scale zonation in Australian saltmarsh vegetation remain poorly understood. There is also a paucity of studies on the tolerance of Australian saltmarsh plants to variations in soils, with implications for conservation and restoration (Adam 2009).

Australia, by global standards, has only a small extent of its coastline occupied by saltmarshes: approximately 16,000 km^2 (Saintilan & Adam 2009). They are generally limited to the southeast of the continent, including Tasmania, with small areas in the southwest of Western Australia (Chapman 1974, Saintilan 2009a). Fairweather (1990) noted that Australian saltmarshes had received the least attention of all marine habitats and their ecological values were largely ignored. Nevertheless, during the past decade a growing appreciation of the value of saltmarshes and a realisation that predicted climate change and sea-level rise will impact saltmarshes directly, has led to increased research in this challenging environment (Saintilan 2009b). This has culminated in the "Subtropical and Temperate Coastal Saltmarsh Community" being recently listed in the 'vulnerable' category of the Australian Government's *Environment Protection and Biodiversity Conservation Act 1999* (Threatened Species Scientific Committee 2013).

Tasmania has about 58.6 km^2 of coastal saltmarshes, principally located on the east coast and Flinders Island, the north coast and the far northwest including King Island (Prahalad & Kirkpatrick 2019). Although only a small proportion of the Australian total, Tasmania features the highest diversity of saltmarsh vegetation communities

(Saintilan 2009a, 2009b). Pioneering Tasmanian saltmarsh research was conducted by Curtis & Somerville (1947) at Boomer Bay on the Tasman Peninsula. Other early work focused on intertidal ecology, principally with respect to algae (Guiler 1949, 1952), and the distribution, mapping and vegetation of saltmarshes (Kirkpatrick & Glasby 1981). Work on the benthic fauna, vegetation and soil factors continued sporadically in the 1980s and 1990s (Wong *et al.* 1993, Richardson & Mulcahy 1996, Richardson *et al.* 1997, 1998). Gouldthorpe (2000) researched the impacts of drainage and grazing on Derwent River marshes and a recent study identified changes in the extent and community composition of southeast Tasmanian saltmarshes (Prahalad *et al.* 2011). The real and projected impacts of climate change have also received recent attention (Mount *et al.* 2010, Prahalad *et al.* 2015, Prahalad & Kirkpatrick 2019). Nevertheless, there are no Tasmanian studies with detailed field measurements that focus on the interactions of saltmarsh soils and vegetation.

The present study aims to redress this shortcoming by investigating and detailing the relationships between vegetation and a broad array of edaphic factors along a saltmarsh to woodland gradient at a site in eastern Tasmania. Specifically, we ask: (i) what vegetation communities are present in our study site as identified by classification methods?; (ii) what local edaphic factors are likely to influence the presence and distribution of identified vegetation communities?; (iii) how do edaphic factors vary between saltmarsh and the adjacent woodland zone?; (iv) how do edaphic factors vary with elevation and reflect the vegetation community composition along a saltmarsh to woodland gradient? We therefore endeavour to provide a deeper understanding of the edaphic factors that require consideration from a coastal saltmarsh conservation and restoration perspective.

METHODS

Study location

Long Point is on the western shore of Moulting Lagoon, an internationally listed Ramsar site located in eastern Tasmania (fig. 1; Prahalad & Kriwoken 2010). The Lagoon is a wave-dominated estuary with a low-energy central basin that is rimmed by expansive intertidal environments and a coastal parallel barrier (Harris *et al.* 2002, Heap *et al.* 2004). The tidal regime is semi-diurnal with a microtidal range of 1.2 metres. The current mean annual rainfall is 590 mm, the mean annual maximum temperature is 18.5 °C and the mean annual minimum temperature is 9.4 °C. Long Point occupies a largely intertidal area of 386 ha made up of the largest contiguous saltmarsh patch in Tasmania (359 ha; Prahalad & Kirkpatrick 2019); the balance is coastal grassland and woodland. The land is privately owned by the Tasmanian Land Conservancy (TLC) and managed for conservation purposes (Prahalad & Kriwoken 2010). The peninsular site is bisected by a central stabilised sand dune varying from 5 to 20 m in elevation which divides the saltmarsh into western and eastern sections. A dolerite ridge defines the extreme southern margin of the site. Fluvial processes have played a major role in the formation of Long Point's topographic framework with the underlying grey silts deposited during the Holocene (Kiernan 2013).

The site is characterised by extensive, low-lying saltmarsh dominated by succulent herbs and shrubs, primarily *Sarcocornia* spp. along with *Disphyma crassifolium* and *Tecticornia arbuscula*. The saline graminoids are dominated by *Gahnia filum* and *Austrostipa stipoides*. The lowland grasslands and woodland contain native (e.g., *Pteridium esculentum*), and introduced (e.g., *Ulex europaeus*) plants and with overstorey species such as *Acacia mearnsii* in the woodland areas (Kingdom 2008). Vegetation communities on the site, particularly the saltmarsh section, are generally well-defined. Here, the saltmarsh vegetation has been broadly classified under the Tasmanian Vegetation Monitoring and Mapping Program (TASVEG 3.0, version 2013) as ASS (saline succulent herbland), ARS (saline sedgeland/rushland). The grassland and woodland vegetation are classified as GPL (lowland *Poa* grassland) and NBA (*Bursaria–Acacia* woodland/grassland), and the sand dune classified as DVG (*Eucalyptus viminalis* woodland) and FRG (regenerating cleared land) (Harris & Kitchener 2005, Department of Primary Industries, Parks, Water and Environment 2014). The succulent herbland and saline

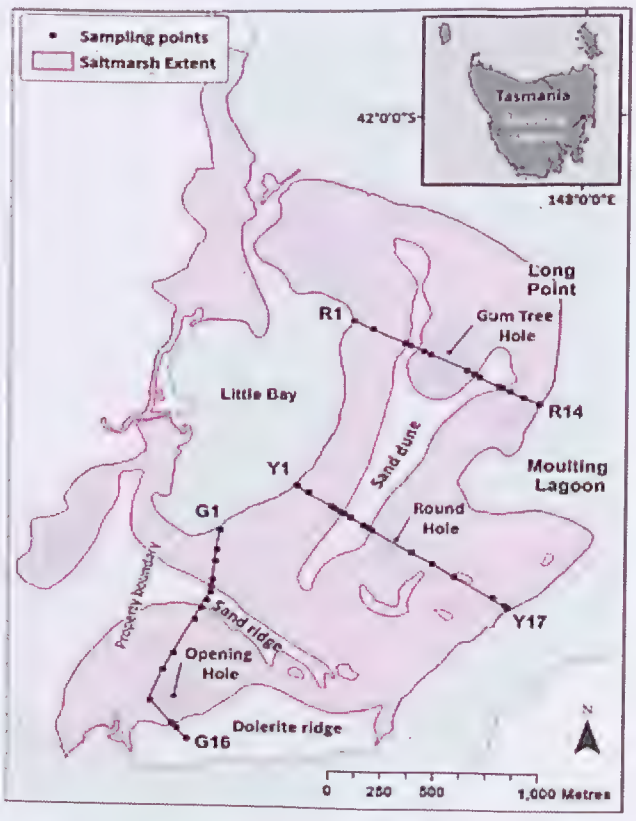


FIG. 1 — Long Point study location with key physical and geomorphological features noted. The transect lines R, Y and G were derived from the 47 individual sampling points. The saltmarsh extent is denoted by the polygon and the woodland areas occupy the sand dune, sand ridge and the dolerite ridge. Base data from the LIST, © State of Tasmania.

grasslands were used for sheep grazing prior to acquisition by the TLC in 2005, resulting in the presence of a number of introduced plant species.

Sampling design

Sampling of vegetation and soil was conducted along three transects established across Long Point. Two transects (R and Y) were orientated west to east and traversed the sand dune. The third transect (G) tracked in a southerly direction incorporating the sand ridge and ended on the dolerite ridge. Transect locations and direction were determined by the profile of the landform and originally positioned to capture as much environmental variation as possible across the elevation gradient (table 1, also see fig. 1).

The boundaries of vegetation communities at Long Point are visually discernible based on the dominance of two or three species within each community and this was used as the basis for allocating individual sampling points along each transect. Sampling points were positioned towards the centre of each vegetation community to avoid the influence of edge effects. Along each transect, a sampling point was located every time the vegetation community composition changed. This allowed for an increased sample size with multiple samples collected from similar vegetation communities across the study area. At each sampling point, a modified Braun–Blanquet cover-abundance method was applied with a 1×5 m quadrat at right angles to the transect to estimate the quantity of each species in a vegetation community in a single scale which incorporates both cover and abundance (Mueller-Dombois & Ellenberg 1974, Moore & Chapman 1986). This method ascribes an ordered numerical value to the crown cover percentage of individual species as follows:
1 = < 1%, 2 = 1–5%, 3 = 5–25%, 4 = 25–50%, 5 = 50–75% and 6 = > 75%

Due to the overlapping cover of some species, the total cover for each sample plot may exceed 100% (Moore & Chapman 1986) and this was a common outcome at Long Point. The nomenclature, classification and current status (endemic, introduced etc.) of plant species conforms to de Salas & Baker (2018). All sampling points (n = 47) were assessed in April 2013 and reassessed in January 2014 to account for summer annuals.

Three replicate soil samples to a depth of 10 cm were taken at each sampling point. The sampling depth was chosen to include the root zone. The summer soil sampling was conducted in January 2013, with a follow-up winter soil sampling in July 2013. The winter soil sampling regime was conducted to document changes in soil moisture content, pH and EC due to seasonal changes in precipitation and evaporation at the site.

At each sampling point, surface elevation was measured using a real-time kinematic survey with geodetic grade GPS receivers. A base station was set up on a 4th order Survey Control Point located on Grange Hill near Long Point using a Leica 1200 GPS and a radio transmitter. A Leica 1200 Rover GPS was used to measure each sampling point (n = 47) at 1 second epochs for 20 seconds. The accuracy of these points relative to the base station was ±20 mm horizontally and ±50 mm vertically.

Laboratory analyses

Soil from each sample was air-dried in a fume hood using only fan-forced air to prevent mould growth or any changes to the characterisation of the soil. Once dried, the samples, minus any obvious plant material, were passed through a 2 mm sieve. The dry-sieved fraction was packaged in seal-top plastic bags, labelled and stored. The analysis of the summer and winter soil samples for moisture content and soil chemistry (pH and EC), and of the combined samples for soil organic matter, followed Rayment & Lyons (2011). As salinity and EC have a close relationship (Hazelton & Murphy 2007), EC was used as a proxy for salinity in this study. The analysis for total carbon was conducted using an ELTRA® CS 2000 Carbon Sulfur Determinator. The standard procedure for carbon analysis in the ELTRA, as detailed in the operating manual, was followed. A particle size analysis (PSA) was carried out on each of the combined soil samples to classify the soil texture at each sampling point. The analysis was based on the density method which involves the Bouyoucos hydrometer procedure (Day 1965, Sur & Kukal 1992, Loveland & Whalley 2000).

TABLE 1 — Transect attributes at Long Point, Moulting Lagoon, Tasmania

Transect	Description	Length (m)	Sampling points (n)
R	Northern transect through Gum Tree Hole which crosses the sand dune twice either side of the lunette; sand dune has a woodland-dominated vegetation cover. Saltmarsh vegetation is extensive on either side of the sand dune and around Gum Tree Hole.	995	14
Y	Middle transect through Round Hole after crossing the sand dune; vegetation either side of the dune is dominated by graminoids (some introduced). Saltmarsh vegetation is extensive either side of the sand dune and around Round Hole.	1215	17
G	Southern transect which crosses the low east/west sand ridge and is dominated by bracken; it traverses beside Opening Hole and ends on the dolerite ridge. This transect had two unavoidable bends due to impenetrable gorse cover present at the time of establishment.	1320	16
	Totals	3530	47

Statistical analyses

To characterise the distinct vegetation communities present in the study area, vegetation data from the 47 sampling points were grouped by cluster analysis incorporating the Ward linkage method from the *vegan* package in R (Oksanen 2013). The analysis used data trimmed to cover-abundance values of 3–6 to distinguish vegetation groups. Cover-abundance values of 1 and 2 (i.e., values < 5%) were excluded as they unduly confounded the cluster analysis (Wildi 2013). The preferred grouping of the data was determined through a whole-evidence approach that included a statistical analysis of cover data, a visual appraisal and first-hand knowledge of the site (Wildi 2013). Non-metric multidimensional scaling (nMDS) was then used to explore the relationship between the sampling points and the six groups. The standard TASVEG 3.0 vegetation community groups for saltmarsh were used to relate to, and compare with, the six groups derived for this study.

Indicator species emerge through an analysis of occurrence or abundance values from a set of sampled points and the classification of the same points into site groups, which may represent vegetation communities. The indicator species analysis was carried out in the R package *indicspecies* using the indicator value function (indval) (De Cáceres 2013).

Data from edaphic factors were plotted against the elevation profile of each transect to display broad response patterns. The mean values from the summer and winter analysis of pH, EC and moisture were plotted to emphasise seasonal differences, while the outcome of SOM, carbon and the texture elements were plotted as percentages.

Soil data from each sampling point were aligned to the vegetation groups — ASS(a), ASS(b), ARS, GSL (lowland grassy sedgeland), GPL(sr) and GPL(dr) — using multivariate methods in the *vegan* package in R. Boxplots were used to summarise the spread of the data differences between group means and were examined using analysis of variance (ANOVA). The post hoc Tukey's Honestly Significant Difference (HSD) test was used to identify those groups that differed significantly from each other for each soil factor. The relationship between edaphic factors was tested using the correlation function in R to help identify predictor variables that would be useful tools in the future. Soil texture analysis was carried out in the R package *soiltexture* (Moeys 2014). The results of PSA for each sample along with the average for each vegetation group were plotted on the standard USDA/FAO triangular texture diagram as recommended by Minasny & McBratney (2001), to summarise the soil texture classification for each sampling point.

RESULTS

Vegetation communities

Cluster analysis of the vegetation data identified six distinct vegetation communities present in the study area (fig. 2). Based on the dendrogram of the three defined saltmarsh vegetation communities, two groups at Long Point fell into the saline succulent herbland (ASS) code, one that was wholly dominated by *Sarcocornia quinqueflora* and/or *S. blackiana*, the other dominated by *T. arbuscula*, *S. quinqueflora*

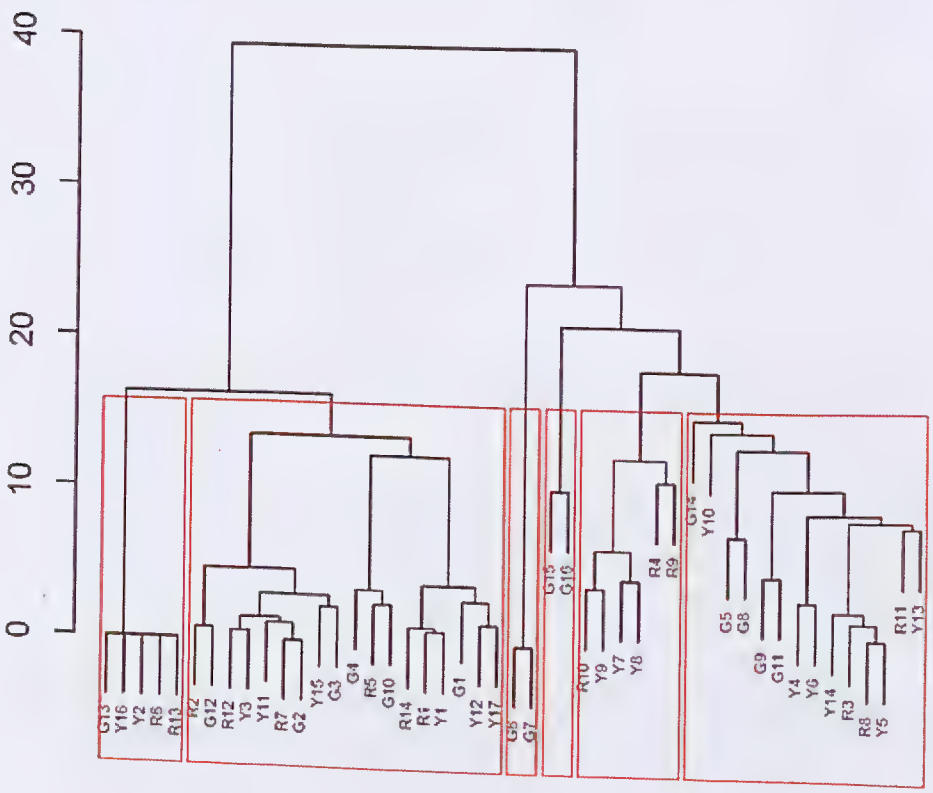


FIG. 2 — Dendrogram of 47 sampling points from transects R, Y and G (see fig. 1) clustered by Ward linkage method into six groups based on vegetation communities differentiated to a level of 16% dissimilarity. Groups have been assigned TASVEG 3.0 codes (see table 2 for group descriptions) as follows: (named from left to right) ASS(a), ASS(b), GPL(sr), GPL(dr), GSL and ARS.

and/or *S. blackiana* and *D. crassifolium*. These two groups were annotated ASS(a) and ASS(b) respectively. The split in ASS was justified by vegetation structure (height) as observed on the ground. The third group represented saline sedgeland/rushland dominated by *Juncus kraussii* and *G. filum* which is consistent with the TASVEG community coded as ARS.

The ecotone zones and woodland mapped as GSL were dominated by *Lomandra longifolia* and to a lesser extent *Lepidosperma concavum*, with tall shrubs such as *A. mearnsii* and *Bursaria spinosa*. Lowland grasslands, represented by code GPL, were split into two groups (fig. 2), GPL(sr) and GPL(dr), to represent the on-ground variation noted in the vegetation community. GPL(sr) captured vegetation dominated by *Pteridium esculentum* and *J. kraussii*, while GPL(dr) included areas dominated by tussocks of *Poa labillardierei*. Group descriptions are provided in table 2.

The results of the indicator species analysis are listed in table 3 highlighting vegetation taxa for which $p < 0.05$. At Long Point, *D. crassifolium* was a significant indicator species ($p < 0.02$) in all three saltmarsh groups. Similarly, *S. quinqueflora* was highly significant in groups ASS(a) and ASS(b) ($p < 0.001$) but was also evident in ARS as a groundcover species, although in much reduced extent. Similarly, *A. stipoides* was found on the sand dune (GSL) and dolerite ridge (GPL(dr)) as well as the saline grasslands (ARS) ($p < 0.015$).

Soil characteristics and edaphic relationships

Soil data for each transect are summarised in figure 3A–C. The elevation measured across the study area ranged from 0.21 m to greater than 10 m above mean sea level (see lowest panel in fig. 3A–C). Within the saltmarsh, elevation ranged from 0.21 m to 0.84 m. The elevation of the woodland zone ranged from 1 m to greater than 10 m. Summer moisture and EC, winter moisture and EC, carbon and SOM were all strongly inversely related to elevation. Small changes in elevation had a marked impact on moisture and EC. Similarly, although to a lesser extent, summer and winter pH were negatively correlated with elevation. The response of pH may be aligned more to a combination of elevation and vegetation community structure. Texture was affected by landscape position, including elevation. Sand content was positively correlated with elevation. In contrast, silt and clay content were negatively related to elevation and sand content. The strength of the correlation (table 4) between each variable showed that SOM and winter moisture have the highest correlation with the remaining edaphic factors. SOM shows very little correlation with pH in summer and winter, but strong correlation with the remaining factors. Winter moisture displayed moderate correlation with summer and winter pH, with strong correlation with the remaining factors.

Following the alignment of the sampling plots to vegetation groups, results for each edaphic factor were summarised in boxplots by vegetation group (fig. 4A–H, also see table 5 for ANOVA outputs). Soil moisture varied 40-fold from 2.4% (summer GPL(dr)) to a high of 92.5%. The group medians for moisture (fig. 4G, H) were significantly different between each of the saline vegetation groups (ASS(a), ASS(b) and ARS); yet not significantly different between the groups on non-saline soils (GSL, GPL(sr) and GPL(dr)). Soil pH (fig. 4C, D) ranged from 3.74 (summer GPL(sr)) to 7.48 (winter ASS(b)); group medians in ASS(a) and ARS displayed similarity for both summer and winter. GSL and GPL(dr) were similar in summer, yet dissimilar in winter. Between saltmarsh groups, the pH of ASS(a) was dissimilar to the other two groups.

EC varied by over 2000-fold from 0.02 dS/m (summer GPL groups) to a high of 49.6 dS/m (winter ASS(a)). Group medians for saline soils (fig. 4E, F) displayed a level of dissimilarity for both summer and winter, reflecting varying degrees of marine water inundation. There was very little difference in the non-saline soils for both seasons. SOM varied 25-fold from less than 2% (GSL) to a high of over 50% (ASS(b)). Group medians for SOM (fig. 4G) were dissimilar for saline soils with some overlap between ASS(b) and ARS, with similarity for the non-saline soils. In respect of carbon (fig. 4H), all groups except for ASS(a) displayed similarity. ANOVA outputs of all factors are presented in table 5. Tukey HSD results are presented in table 6.

TABLE 2 — Assigned vegetation community codes at Long Point, Moulting Lagoon, Tasmania

Sampling points	TASVEG Code	Assigned Code	Description
5	ASS	ASS(a)	Succulent saline herbland wholly dominated by *Sarcocornia quinqueflora* and/or *S. blackiana.*
18	ASS	ASS(b)	Succulent saline shrubland dominated by bare areas, *Tecticornia arbuscula, S. quinqueflora* and/or *S. blackiana,* and *Disphyma crassifolium.*
14	ARS	ARS	Saline sedgeland/rushland dominated by *Juncus kraussii, Gahnia filum, Austrostipa stipoides* and, to a lesser extent, *Poa labillardierei.*
6	GSL	GSL	Dominated by graminoid *Lomandra longifolia* with *Acacia mearnsii* and *Bursaria spinosa* tall shrubs interspersed with lowland grasslands.
2	GPL	GPL(sr)	Lowland grassland dominated by *Pteridium esculentum* and *J. kraussii.*
2	GPL	GPL(dr)	Lowland grassland on dolerite soils dominated by *P. labillardierei* and low or prostrate herbs.

TABLE 3 — Indicator vegetation species for each vegetation community ($p < 0.05$) ordered by indicator value (ind val)

Group	Vegetation species	ind val	p.value
ASS(a) (succulent herbland – a)	*Sarcocornia quinqueflora*	0.944	0.001
	Disphyma crassifolium	0.822	0.011
ASS(b) (succulent shrubland – b)	Bare ground	0.961	0.002
	Sarcocornia quinqueflora	0.944	0.001
	Tecticornia arbuscula	0.904	0.001
	Disphyma crassifolium	0.822	0.011
ARS (saline grassland)	Bare ground	0.961	0.002
	Poa labillardierei	0.939	0.001
	Juncus kraussii	0.869	0.002
	Gahnia spp.	0.854	0.001
	Disphyma crassifolium	0.822	0.011
	Austrostipa stipoides	0.807	0.015
GSL (sand dune)	*Ehrharta stipoides*	1.000	0.001
	Bare ground	0.961	0.002
	Oxalis perennans	0.894	0.001
	Lomandra longifolia	0.877	0.005
	Austrostipa stipoides	0.807	0.015
	Leontodon saxatilis[1]	0.764	0.017
	Poa rodwayi	0.707	0.011
GPL(sr)	*Hibbertia prostrata*	1.000	0.003
	Bare ground	0.961	0.002
	Poa labillardierei	0.939	0.001
	Ficinia nodosa	0.935	0.002
	Oxalis perennans	0.894	0.001
	Lomandra longifolia	0.877	0.005
	Pteridium esculentum	0.835	0.018
	Isolepis nodosa	0.800	0.027
GPL(dr)	*Aira caryophyllea*	1.000	0.005
	Austrodanthonia setacea	1.000	0.005
	Bare ground	0.961	0.002
	Poa labillardierei	0.939	0.001
	Juncus kraussii	0.869	0.002
	Zoysia macrantha	0.910	0.002
	Oxalis perennans	0.894	0.001
	Lomandra longifolia	0.877	0.005
	Austrostipa stipoides	0.807	0.015
	Baumea juncea	0.801	0.022
	Poa rodwayi	0.707	0.011

[1] Introduced species

The soil texture triangle (fig. 5) demonstrates the decrease in clay content and increase in sand content from the lower marsh to the upper marsh, with further increases of sand content into the woodland indicating the influence of the local sand dune and ridge.

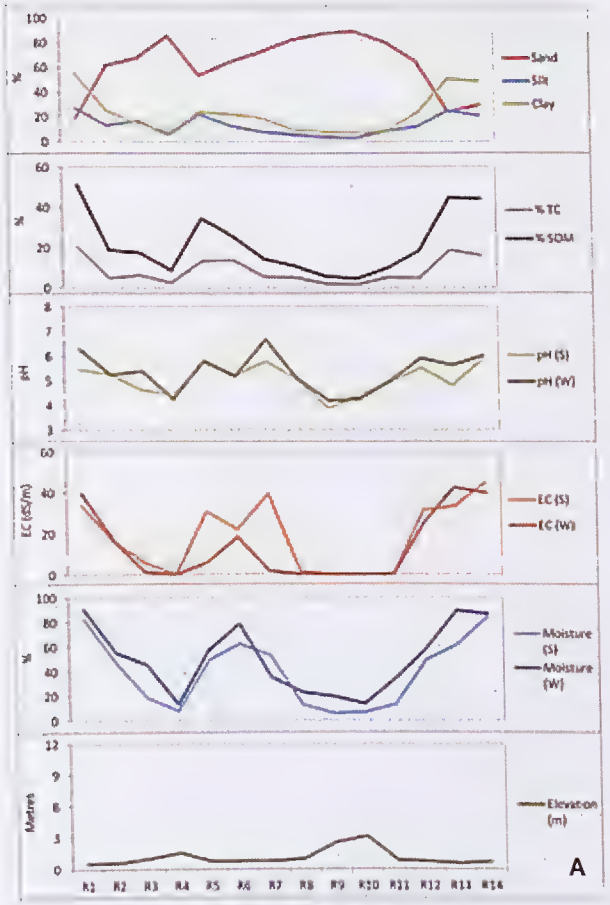
DISCUSSION

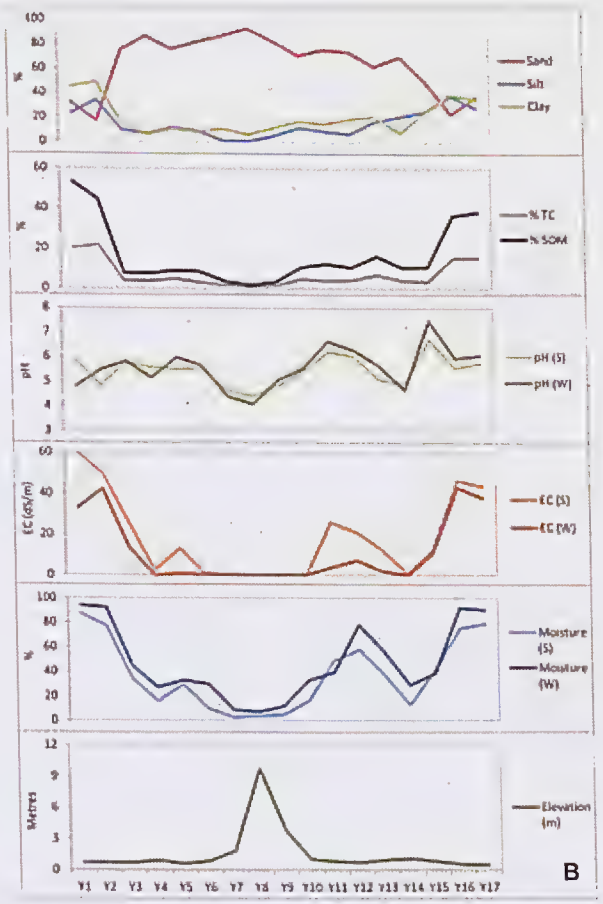
Our study objectively identified three saltmarsh vegetation groups within Long Point based on evidence from vegetation structure, composition and position in the landscape. This extends the findings of other comparable state-wide assessments by Richardson *et al.* (1997, 1998), who subjectively recognised three broad vegetation groups within Tasmanian saltmarshes coinciding with low-, mid- and high-marsh areas. The succulent saline herbland (the low marsh) is largely described by *S. quinqueflora*, the succulent saline shrubland (mid marsh) characterised by *T. arbuscula*, and the saline grassland (upper marsh) by *G. filum*, *J. kraussii* and other graminoids (see table 3). The classification of saltmarsh vegetation communities in our study is broadly consistent with Adam's (1990) summary of southern Australian saltmarshes, which recognised three vegetation categories based on dominant growth form, including herb communities, dwarf shrubs, and graminoids.

Our three saltmarsh and three grassy woodland groups are largely consistent with the TASVEG classification (see table 2). However, consideration should be given to subdividing succulent herbland (ASS) into two communities, one dominated by *Sarcocornia* spp., the other by *T. arbuscula*, for ecological studies.

Apart from a limited number of studies (e.g., Clarke & Hannon 1967, Richardson & Mulcahy 1996), consideration of environmental gradients in the saltmarsh to woodland zone has not been common. Soil conditions at our study area were clearly aligned with the vegetation groups. This was not unexpected between the saltmarsh and woodland groups, but notable differences also existed between the saltmarsh groups. Our analysis showed that the key predictors were winter moisture and SOM, with winter moisture being the better indicator. As a predictor, SOM is more difficult to measure as it requires laboratory testing with highly accurate weighing equipment and a 550°C muffle furnace (Rayment & Lyons 2011), whereas winter moisture can be measured in the field with an inexpensive handheld meter.

Although elevation showed only minor variation in the saltmarsh zone (a range of 0.306 m), it clearly plays an important role in the saltmarsh environment through its interaction with tidal flows. Typically, the saltmarsh zone will gently rise in height from the seaward fringe and tidal creeks to its landward limit or the terrestrial zone (Long & Mason 1983). This elevational gradient determines the temporal pattern of inundation by tidal flooding (Sánchez *et al.* 1996), and results in decreasing tidal influence as elevation increases (Long & Mason 1983). This pattern was apparent in elevation data at Long Point. The elevation range of the three saltmarsh vegetation groups began at





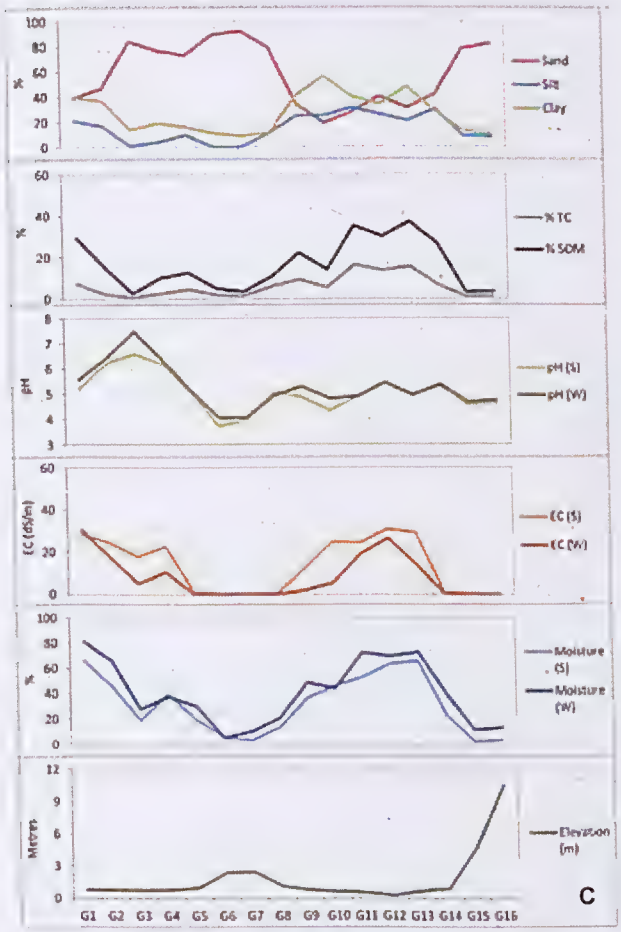


FIG. 3 — (**A**) Results of soil analysis of transect R plotted with elevation profile (plots are not equally spaced in the field); note that all factors except for sand (top plot) are aligned inversely to elevation. R1 = Little Bay; R14 = Moulting Lagoon; R4 and R10 – summits of sand dune; between R6 and R7 – Gum Tree Hole (lunette). (**B**) — Results of soil analysis of transect Y plotted over elevation profile (plots are not equally spaced in the field), note that all factors except for sand (top plot) are aligned inversely to elevation. Y1 = Little Bay; Y17 = Moulting Lagoon; Y8 – summit of sand dune; between Y12 and Y13 – Round Hole (lunette); Y4 and Y14 are slightly higher in elevation than surrounding marsh land: note the change in moisture, EC and pH. (**C**) — Results of soil analysis of transect G plotted over elevation profile (plots are not equally spaced in the field); note that all factors except for sand (top plot) are aligned inversely to elevation. G1 = Little Bay; G16 = summit of dolerite ridge; G6 and G7 = summit of sand ridge; between G12 and G13 = Opening Hole. Legend note: TC (total carbon) by ELTRA = dry combustion method, SOM (organic carbon) by loss on ignition, EC = electrical conductivity, S = summer, W = winter.

TABLE 4 — Correlation between each edaphic factor

Edaphic factor	SOM	Total carbon	pH (S)	pH (W)	EC (S)	EC (W)	Moist (S)	Moist (W)
SOM	1.000[1]	0.965	0.225	0.211	0.804	0.847	0.873	0.894
TC	0.965	1.000	0.154	0.150	0.768	0.810	0.830	0.858
pH (S)	0.225	0.154	1.000	0.891	0.478	0.294	0.483	0.413
pH (W)	0.211	0.150	0.891	1.000	0.471	0.319	0.480	0.415
EC (S)	0.804	0.768	0.478	0.471	1.000	0.846	0.940	0.861
EC (W)	0.847	0.810	0.294	0.319	0.846	1.000	0.867	0.880
Moist (S)	0.873	0.830	0.483	0.480	0.940	0.867	1.000	0.950
Moist (W)	0.894	0.858	0.413	0.415	0.861	0.880	0.950	1.000

[1] 0 = no correlation, 1.0 = the highest level of correlation
[2] TC = total carbon, EC = electrical conductivity, S = summer, W = winter, Moist = water content.

TABLE 5 — ANOVA output of all edaphic factors ordered by decreasing F value

Soil variable	Df[1]	F value	p.value
Moisture (S)	5,41	24.31	<0.001
EC (S)	5,41	21.90	<0.001
Moisture (W)	5,41	17.97	<0.001
pH (S)	5,41	16.08	<0.001
pH (W)	5,41	12.50	<0.001
EC (W)	5,41	11.01	<0.001
Total carbon	5,41	7.65	<0.001
SOM	5,41	6.97	<0.001

[1] Df = degrees of freedom.

0.256 m in ASS(a), through to 0.300 m in ASS(b) and reached a maximum of 0.835 m in ARS. These values mirror a study in northwest Spain where Sánchez *et al.* (1996) found the levels separating the low marsh from the mid marsh to the high marsh were 0.260 m, 0.350 m to 0.400 m respectively. In both studies, elevation was referenced to mean sea level.

Zonation, as discussed above, is linked to tidal inundation which is determined by elevation (Sánchez *et al.* 1996, Huckle *et al.* 2000). Plant distribution is therefore not a direct reflection of elevation within the saltmarsh; rather, it is more a consequence of inundation controlled by elevation (Long & Mason 1983). Some plant species are limited to precise elevational classes. However, many plants appear in two zones and some appear in all three (Sánchez *et al.* 1996). In an Alaskan saltmarsh, Vince & Snow (1984) found that vegetation communities changed markedly with increasing elevation, sometimes over an elevation difference of less than 8 cm with several individual species ranging over a number of zones. Inundation frequency therefore has a greater explanatory value for forecasting the occurrence of dominant species than has altitude (Davis & Gray 1966, Bockelmann *et al.* 2002). Minor changes in elevation, particularly closer to sea level, lead to greater variations in inundation, which in turn result in greater variation in vegetation. However, some plant species can still be found over a wide range of inundation frequencies (Bockelmann *et al.* 2002). There is no simple explanation as to why some species occupy two or more zones, except that some individual plant species may be more adapted to, or tolerant of, a wide range of saltmarsh conditions. Also, of potential consequence is interspecific competition between species (Snow & Vince 1984, Pennings & Callaway 1992).

Edaphic factors generally show gradients from the low marsh to the high marsh that correspond with elevation and tidal impacts (Pennings & Callaway 1992). Throughout the study site, clear patterns of correlations among the edaphic factors were evident. Moisture and EC (summer and winter) along with SOM and carbon, all showed a distinctive negative correlation with elevation. The pH (summer and winter), though less distinctive, was still negatively correlated with elevation.

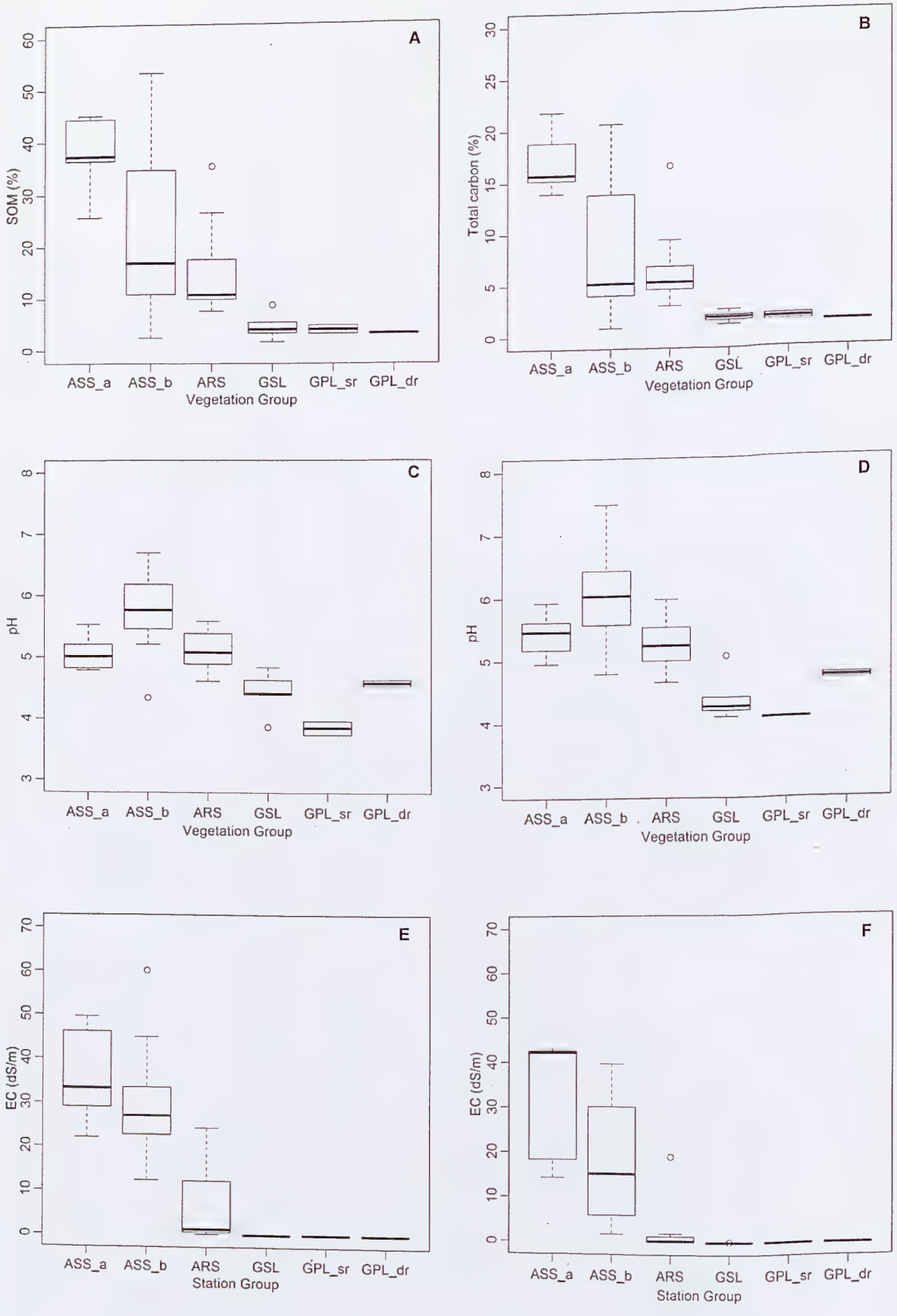
Saltmarsh areas that are prone to frequent inundation have high levels of moisture, whereas areas of reduced inundation frequency record lower levels of moisture content (Long & Mason 1983). The sampling points adjacent to the coastal margin and to the ephemeral ponds recorded levels of moisture exceeding 61% in summer and 72% in winter, with group ASS(a) recording means of 68% and 85% respectively. Decreasing moisture content was strongly evident progressing through the middle marsh to the upper marsh with means of 55% and 22% respectively for summer and 60% and 37% for winter. Similarly, Richardson & Mulcahy (1996) reported high levels of moisture throughout the saltmarsh zone on Bruny Island with levels decreasing on approach to the woodland zone.

Decreasing tidal inundation as a result of increasing elevation of the saltmarsh zone most often results in a decrease in soil salinity (Adam 1990, Silvestri *et al.* 2005). Salinity levels in the low marsh are relatively constant throughout the year as tidal inundation recharges the water table and the soils (Adam 1990). However, the middle and upper marsh zones are subject to seasonal and temporal

TABLE 6 — Group means, standard error, range and Tukey groups for each edaphic factor[1]

Variable	Group	*n*	Mean ± Std Error	Min	Max	Tukey Groups
Moist (S)	ASS(a)	5	68.4 ± 3.3	61.59	77.12	a
	ASS(b)	18	55.2 ± 4.4	18.98	87.31	a
	ARS	14	22.4 ± 3.3	10.00	53.33	b
	GSL	6	5.3 ± 0.8	2.59	7.87	b
	GPL(sr)	2	4.6 ± 1.4	3.29	6.01	b
	GPL(dr)	2	3.0 ± 0.6	2.38	3.53	b
Moist (W)	ASS(a)	5	85.1 ± 4.0	72.89	92.45	a
	ASS(b)	18	60.8 ± 5.2	27.79	94.25	a
	ARS	14	37.5 ± 3.8	21.20	72.41	b
	GSL	6	12.5 ± 1.7	7.44	19.26	c
	GPL(sr)	2	7.9 ± 2.4	5.46	10.40	c
	GPL(dr)	2	12.4 ± 0.9	11.46	13.33	c
pH (S)	ASS(a)	5	5.1 ± 0.1	4.79	5.53	b c
	ASS(b)	18	5.8 ± 0.1	4.34	6.70	a
	ARS	14	5.1 ± 0.1	4.61	5.58	b
	GSL	6	4.4 ± 0.1	3.87	4.48	c d
	GPL(sr)	2	3.9 ± 0.1	3.74	3.97	d
	GPL(dr)	2	4.6 ± 0.0	4.54	4.63	b c d
pH (W)	ASS(a)	5	5.4 ± 0.2	4.95	5.93	a b
	ASS(b)	18	6.1 ± 0.2	4.79	7.48	a
	ARS	14	5.3 ± 0.1	4.65	5.97	b
	GSL	6	4.4 ± 0.1	4.08	5.05	c
	GPL(sr)	2	4.1 ± 0.0	4.07	4.07	c
	GPL(dr)	2	4.7 ± 0.0	4.67	4.76	b c
EC (S)	ASS(a)	5	36.1 ± 5.2	22.18	49.64	a
	ASS(b)	18	29.7 ± 2.7	12.51	60.13	a
	ARS	14	5.5 ± 2.0	0.20	24.48	b
	GSL	6	0.0 ± 0.0	0.03	0.06	b
	GPL(sr)	2	0.1 ± 0.0	0.05	0.08	b
	GPL(dr)	2	0.0 ± 0.0	0.03	0.06	b
EC (W)	ASS(a)	5	32.1 ± 6.5	14.03	43.00	a
	ASS(b)	18	18.4 ± 3.1	1.84	39.86	a
	ARS	14	1.9 ± 1.3	0.08	19.03	b
	GSL	6	0.0 ± 0.0	0.02	0.10	b
	GPL(sr)	2	0.0 ± 0.0	0.02	0.07	b
	GPL(dr)	2	0.0 ± 0.0	0.02	0.02	b
SOM	ASS(a)	5	37.8 ± 3.5	25.66	45.05	a
	ASS(b)	18	23.1 ± 3.7	2.52	53.16	a b
	ARS	14	14.9 ± 2.2	7.69	35.36	b c
	GSL	6	4.7 ± 1.0	1.84	8.86	c
	GPL(sr)	2	4.2 ± 0.8	3.37	4.94	c
	GPL(dr)	2	3.4 ± 0.1	3.30	3.57	c
Total carbon	ASS(a)	5	17.0 ± 1.4	13.85	21.70	a
	ASS(b)	18	8.2 ± 1.5	0.78	20.48	b
	ARS	14	6.2 ± 0.9	2.90	16.38	b
	GSL	6	1.7 ± 0.2	1.04	2.52	b
	GPL(sr)	2	1.7 ± 0.3	1.38	1.97	b
	GPL(dr)	2	1.3 ± 0.1	1.21	1.34	b

[1] Within each edaphic factor, the values followed by the same letter are not different at $p < 0.05$.


A
SOM (%)
0
10
20
30
40
50
60
ASS_a
ASS_b
ARS
GSL
GPL_sr
GPL_dr
Vegetation Group
B
Total carbon (%)
0
5
10
15
20
25
30
ASS_a
ASS_b
ARS
GSL
GPL_sr
GPL_dr
Vegetation Group
C
pH
3
4
5
6
7
8
ASS_a
ASS_b
ARS
GSL
GPL_sr
GPL_dr
Vegetation Group
D
pH
3
4
5
6
7
8
ASS_a
ASS_b
ARS
GSL
GPL_sr
GPL_dr
Vegetation Group
E
EC (dS/m)
0
10
20
30
40
50
60
70
ASS_a
ASS_b
ARS
GSL
GPL_sr
GPL_dr
Station Group
F
EC (dS/m)
0
10
20
30
40
50
60
70
ASS_a
ASS_b
ARS
GSL
GPL_sr
GPL_dr
Station Group

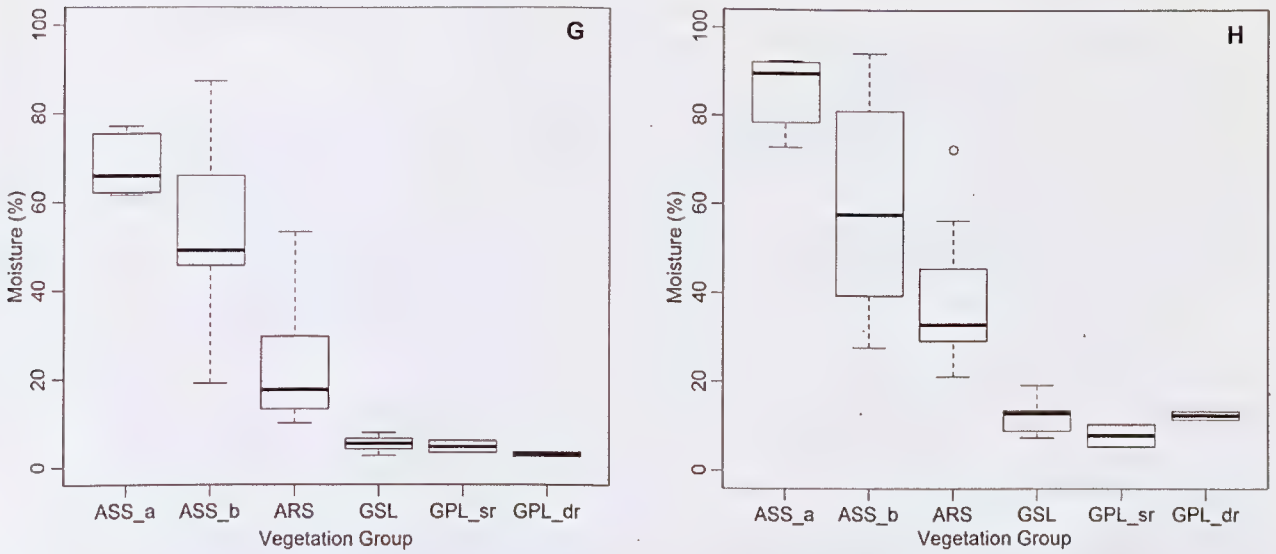


FIG. 4 — (**A**) SOM (soil organic matter) by vegetation group. (**B**) Total carbon by vegetation group.(**C**) Summer pH by vegetation group. (**D**) Winter pH by vegetation group. (**E**) Summer EC by vegetation group. (**F**) Winter EC by vegetation group. (**G**) Summer moisture by vegetation group. (**H**) Winter moisture by vegetation group.

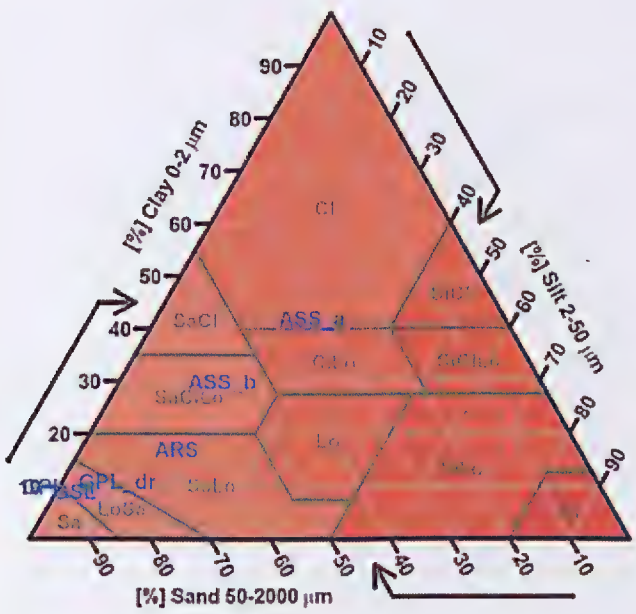


FIG. 5 — USDA/FAO soil texture triangle with data for clay, silt and sand plotted by vegetation group (marked in blue). Soil classifications: ASS_a = Clay; ASS_b = Sandy clay loam; ARS = Sandy loam; GSL, GPL_sr and GPL_dr = Loamy sand. Codes ASS_a, ASS_b and ARS are saltmarsh communities; GSL, GPL_sr and GPL_dr are woodland communities which are clustered in the lower left hand corner of the diagram.

variations in salt levels due to evapotranspiration, which can lead to locally elevated salinity sometimes exceeding that of seawater, but also decreasing salinity following heavy rainfall (Long & Mason 1983, Adam 1990, Álvarez-Rogel *et al.* 1997). Thus, salinity has neither a strictly positive nor negative correlation, but a complex relationship with elevation (Adam 1990). At Long Point, there was a clear decreasing soil EC gradient across the saltmarsh zone, which also decreased from summer to winter. EC values for the ASS(a and b) groups were similar ($p < 0.05$), however that of the ARS group was significantly lower in both seasons. The three groups each recorded lower winter EC readings compared to that of summer following heavy rainfall in July 2013. Sodium (Na^+) levels reported by Richardson & Mulcahy (1996) also declined progressively from the seaward side towards the woodland zone.

Clear patterns in pH values were difficult to determine at Long Point. The means for ASS(a) and ARS were more closely aligned for both summer and winter, with a pH value of 5.1 each in summer, increasing slightly to pH 5.4 and 5.3 respectively during winter. In the ASS(b) group, the means for both summer and winter were lower (summer) and higher (winter) than in the other saltmarsh groups, being pH 4.3 and 6.1 respectively. However, there was a significant change ($p < 0.05$) from the saline grasslands to the adjacent woodland zone.

Levels of organic matter in saltmarsh soils are variable, but typically contents of 8% in the lower marsh, 13% in the middle marsh to 25% in the upper marsh have been reported (Long & Mason 1983). Levels of up to 50% have been recorded in some North American marshes (Adam 1990). There was a consistent pattern of SOM at Long Point. The highest levels were recorded in ASS(a) (37.7±3.5%), followed by decreasing levels in ASS(b) (23.1±3.7%), to ARS (14.9±2.2%) with the transition from saltmarsh to the woodland zone marked by a dramatic

fall in SOM to 4.7±1.0%. A study at Westernport Bay (Victoria, Australia) on a site of *Sclerostegia* (now *Tecticornia*) *arbuscula* and *S. quinqueflora*, similar to ASS(a and b) in this study, estimated values of 50–60% for soil organic matter (Van Der Valk & Attiwill 1983). This value is higher than those recorded at Long Point; however, it does support the notion that the low/middle marsh has a high level of organic matter, which is possibly dependent on the vegetation make-up of the individual zones.

CONCLUSIONS

The most conspicuous characteristic of the saltmarsh area was the distinctive assemblage of plants within each vegetation community and their clear delineation. This facilitated the classification of the saltmarsh into three zones — lower, middle and upper — as widely recognised elsewhere. The composition of the vegetation communities changes with an increase in elevation. The low marsh is a low-diversity community made up of one to three succulent species, whilst the middle marsh community is made up of three or more species, often a blend of succulents from the lower marsh with grasses from the upper marsh, and the upper marsh community is made up of a larger number of species, principally saline graminoids. However, strict species fidelity to certain zones was not evident. Many species can tolerate a range of soil moisture and salinity: *Sarcocornia* spp. and *D. crassifolium* have colonised areas in all three zones. Some saltmarsh species have broadly overlapping salinity tolerance ranges as is evident from the field (Clarke & Hannon 1970, Kirkpatrick & Glasby 1981, Silvestri *et al.* 2005) and in the laboratory (Snow & Vince 1984). Yet saltmarsh zonation, particularly at Long Point, is well-defined. Perhaps complementary roles of tolerance to physical conditions such as moisture and salinity at one end of the gradient, and between-species competition where more species survive at the other end could be the key driver (Snow & Vince 1984, Pennings & Callaway 1992). This knowledge of how edaphic factors relate to coastal saltmarsh vegetation communities and their physical tolerance ranges is useful in the context of the threatened status of saltmarshes and the need for monitoring and recovery actions.

ACKNOWLEDGEMENTS

We thank Denna Kingdom from the Tasmanian Land Conservancy for facilitating use of the Long Point Reserve to carry out this study. Our thanks to John and Jane Cotton for access to the research site through their property. Stuart MacDonald, Colin Sumner, Ross Lucas, Jon Traynor, Christian Mackay and Mel Bottrill gave valuable assistance in the field. Jamie Kirkpatrick assisted with identification of vegetation species. Rob Anders contributed technical assistance with real time GPS survey. Two reviewers provided valuable comments.

REFERENCES

Adam, P. 1990: *Saltmarsh Ecology*. Cambridge University Press, Cambridge: 461 pp.

Adam, P. 2002: Saltmarshes in a time of change. *Environmental Conservation* **29**: 39–61.

Adam, P. 2009: Australian saltmarshes in a global context. *In* Saintilan, N. (ed): *Australian Saltmarsh Ecology*. CSIRO Publishing, Collingwood: 1–21.

Álvarez-Rogel, J., Hernández, J., Ortiz-Silla, R. & Alcaraz-Ariza, F. 1997: Patterns of spatial and temporal variations in soil salinity: Example of a salt marsh in a semiarid climate. *Arid Soil Research and Rehabilitation* **11**: 315–329.

Álvarez-Rogel, J., Alcaraz-Ariza, F. & Ortiz-Silla, R. 2000: Soil salinity and moisture gradients and plant zonation in Mediterranean salt marshes of Southeast Spain. *Wetlands* **20**: 357–372.

Anisfeld, S.C. 2012: Biogeochemical responses to tidal restoration. *In* Roman, C.T. & Burdick, D.M (eds): *Tidal Marsh Restoration*. Island Press, Washington: 39–58.

Bockelmann, A.C., Bakker, J.P., Neuhaus, R. & Lage, J. 2002: The relation between vegetation zonation, elevation and inundation frequency in a Wadden Sea salt marsh. *Aquatic Botany* **73**: 211–221.

Boon, P.I., Allen, T., Carr, G., Frood, D., Harty, C., Mcmahon, A., Mathews, S., Rosengren, N., Sinclair, S., White, M. & Yugovic, J. 2015: Coastal wetlands of Victoria, south-eastern Australia: providing the inventory and condition information needed for their effective management and conservation. *Aquatic Conservation: Marine and Freshwater Ecosystems* **25**: 454–479.

Bridgewater, P. & Cresswell, I. 1999: Biogeography of mangrove and saltmarsh vegetation: implications for conservation and management in Australia. *Mangroves and Salt Marshes* **3**: 117–125.

Bridgewater, P. & Cresswell, I. 2003: Identifying biogeographic patterns in Australian saltmarsh and mangal systems: a phytogeographic analysis. *Phytocoenologia* **33**: 231–250.

Burden, A., Garbutt, R.A., Evans, C.D., Jones, D.L. & Cooper, D.M. 2013: Carbon sequestration and biogeochemical cycling in a saltmarsh subject to coastal managed realignment. *Estuarine, Coastal and Shelf Science* **120**: 12–20.

Chapman, V.J. 1974: *Salt Marshes and Salt Deserts of the World*, second edition. Cramer, Lehre: 392 pp.

Clarke, L.D. & Hannon, N.J. 1967: The mangrove swamp and salt marsh communities of the Sydney district: I. Vegetation, soils and climate. *Journal of Ecology* **55**: 753–771.

Clarke, L.D. & Hannon, N.J. 1969: The mangrove swamp and salt marsh communities of the Sydney district: II. The Holocoenotic complex with particular reference to physiography. *Journal of Ecology* **57**: 213–234.

Clarke, L.D. & Hannon, N.J. 1970: The mangrove swamp and salt marsh communities of the Sydney district: III. Plant growth in relation to salinity and waterlogging. *Journal of Ecology* **58**: 351–369.

Curtis, W. & Somerville, J. 1947: Boomer Marsh–a preliminary botanical and historical survey. *Papers and Proceedings of the Royal Society of Tasmania* **81**: 151–157.

Davis, L.V. & Gray, I.E. 1966: Zonal and seasonal distribution of insects in North Carolina salt marshes. *Ecological Monographs* **36**: 275–295.

Day, P.R. 1965: Particle fractionation and particle size analysis. *In* Black, C.A. (ed): *Methods of Soil Analysis: Part 1: Physical and Mineralogical Properties, including Statisics of Measurement and Sampling*. American Society of Agronomy, Madison: 545–566.

De Cáceres, M. 2013: *How to use the indicspecies package (ver. 1.7.1)*. Centre Tecnològic Forestal de Catalunya, Catalonia, Spain: 29 pp.

de Salas, M.F. & Baker, M.L. 2018: *A Census of the Vascular Plants of Tasmania, including Macquarie Island.* Tasmanian Herbarium, Tasmanian Museum and Art Gallery, Hobart: 156 pp.

Department of Primary Industries, Parks, Water and Environment 2014: *The LIST Maps,* http://maps.thelist.tas.gov.au/listmap/app/list/map (accessed 23 March 2014).

Fairweather, P.G. 1990: Ecological changes due to our use of the coast: research needs versus effort. *Proceedings of the Ecological Society of Australia* **16**: 71–77.

Gouldthorpe, J.J. 2000: The Effects of Drainage and Grazing on Saltmarsh Environments on South-east Tasmania. Unpublished BSc (Honours) thesis, School of Geography and Environmental Studies, University of Tasmania, Hobart.

Guiler, E.R. 1949: The intertidal ecology of Tasmania. *Papers and Proceedings of the Royal Society of Tasmania* **83**: 135–201.

Guiler, E.R. 1952: The nature of intertidal zonation in Tasmania. *Papers and Proceedings of the Royal Society of Tasmania* **86**: 31–62.

Harris, P., Heap, A., Bryce, S., Porter-Smith, R., Ryan, D. & Heggie, D. 2002: Classification of Australian clastic coastal depositional environments based upon a quantitative analysis of wave, tidal, and river power. *Journal of Sedimentary Research* **72**: 858–870.

Harris, S. & Kitchener, A. 2005: *From Forest to Fjaeldmark: Descriptions of Tasmania's Vegetation.* Department of Primary Industries, Parks, Water and Environment, Hobart: 432 pp.

Hazelton, P.A. & Murphy, B.W. 2007: *Interpreting Soil Test Results: What Do All The Numbers Mean?* CSIRO Publishing, Collingwood: 152 pp.

Heap, A.D., Bryce, S. & Ryan, D.A. 2004: Facies evolution of Holocene estuaries and deltas: a large-sample statistical study from Australia. *Sedimentary Geology* **168**: 1–17.

Huckle, J.M., Potter, J.A. & Marrs, R.H. 2000: Influence of environmental factors on the growth and interactions between salt marsh plants: effects of salinity, sediment and waterlogging. *Journal of Ecology* **88**: 492–505.

Kelleway, J.J., Saintilan, N., Macreadie, P.I. & Ralph, P.J. 2016: Sedimentary factors are key predictors of carbon storage in SE Australian saltmarshes. *Ecosystems* **19**: 865–880.

Kiernan, K. 2013: A Preliminary Intrepretation of Landscape Evolution at Long Point (Tasmania). Unpublished report to the School of Geography and Environmental Studies, University of Tasmania, Hobart: 3 pp.

Kingdom, D. 2008: Long Point Moulting Lagoon, Swansea: Reserve Management Plan. Unpublished report to the Tasmanian Land Conservancy, Lower Sandy Bay: 52 pp.

Kirkpatrick, J.B. & Glasby, J. 1981: *Salt Marshes in Tasmania: Distribution, Community Composition and Conservation.* Department of Geography, University of Tasmania, Hobart: 65 pp.

Landi, M. & Angiolini, C. 2015: Soil-plant relationships in Mediterranean salt marshes across dune-cultivated land gradient. *Journal of Coastal Research* **31**: 588–594.

Long, S.P. & Mason, C.F. 1983: *Saltmarsh Ecology.* Blackie & Sons, Glasgow: 160 pp.

Loveland, P.J. & Whalley, W.R. 2000: Particle size analysis. *In* Smith, K.A. & Mullins, C.E. (eds): *Soil and Environmental Analysis.* Marcel Dekker, New York: 281–314.

Minasny, B. & McBratney, A.B. 2001: The Australian soil texture boomerang: a comparison of the Australian and USDA/FAO soil particle-size classification systems. *Australian Journal of Soil Research* **39**: 1443–1451.

Moeys, J. 2014: *The Soil Texture Wizard: R Functions for Plotting, Classifying, Transforming and Exploring Soil Texture Data,* https://cran.r-project.org/web/packages/soiltexture/soiltexture.pdf (accessed 30 June 2014).

Moore, P.D. & Chapman, S.B. 1986: Methods in plant ecology. *In* Goldsmith, F.B., Harrison, C.M. & Morton, A.J. (eds): *Description and Analysis of Vegetation,* second edition. Blackwell Scientific Publications, Oxford: 437–524.

Mount, R.E., Prahalad, V.N., Sharples, C., Tilden, J., Morrison, B., Lacey, M., Ellison, J., Helman, J. & Newton, J. 2010: Circular Head Coastal Foreshore Habitats: Sea Level Rise Vulnerability Assessment: Final Project. Unpublished report to Cradle Coast NRM. Blue Wren Group, School of Geography and Environmental Studies, University of Tasmania, Hobart: 221 pp.

Mueller-Dombois, D. & Ellenberg, H. 1974: *Aims and Methods of Vegetation Ecology.* John Wiley & Sons, New York: 547 pp.

Oksanen, J. 2013: *Multivariate Analysis of Ecological Communities in R: Vegan Tutorial,* http://cc.oulu.fi/~jarioksa/opetus/metodi/vegantutor.pdf (accessed 21 July 2014).

Pennings, S.C. & Callaway, R.M. 1992: Salt marsh plant zonation: the relative importance of competition and physical factors. *Ecology* **73**: 681–690.

Prahalad, V.N. & Kriwoken, L.K. 2010: Implementation of the Ramsar Convention on Wetlands in Tasmania, Australia. *Journal of International Wildlife Law and Policy* **13**: 205–239.

Prahalad, V. & Kirkpatrick, J.B. 2019: Saltmarsh conservation through inventory, biogeographic analysis and predictions of change: case of Tasmania, south-eastern Australia. *Aquatic Conservation: Marine and Freshwater Ecosystems* **29**: 717–731.

Prahalad, V.N., Kirkpatrick, J.B. & Mount, R.E. 2011: Tasmanian coastal saltmarsh community transitions associated with climate change and relative sea level rise 1975–2009. *Australian Journal of Botany* **59**: 741–748.

Prahalad, V., Sharples, C., Kirkpatrick, J. & Mount, R. 2015: Is wind-wave fetch exposure related to soft shoreline change in swell-sheltered situations with low terrestrial sediment input? *Journal of Coastal Conservation* **19**: 23–33.

Rayment, G.E. & Lyons, D.J. 2011: *Soil Chemical Methods – Australasia.* CSIRO Publishing, Collingwood: 495 pp.

Richardson, A.M.M. & Mulcahy, M.E. 1996: The distribution of talitrid amphipods (Crustacea) on a salt marsh in Southern Tasmania, in relation to vegetation and substratum. *Estuarine, Coastal and Shelf Science* **43**: 801–817.

Richardson, A.M.M., Swain, R. & Wong, V. 1997: The crustacean and molluscan fauna of Tasmanian saltmarshes. *Papers and Proceedings of Royal Society of Tasmania* **131**: 21–30.

Richardson, A.M.M., Swain, R. & Wong, V. 1998: Relationship between the crustacean and molluscan assemblages of Tasmanian saltmarshes and the vegetation and soil conditions. *Marine and Freshwater Research* **49**: 785–799.

Rogers, K., Saintilan, N., Howe, A.J. & Rodríguez, J.F. 2013: Sedimentation, elevation and marsh evolution in a southeastern Australian estuary during changing climatic conditions. *Estuarine, Coastal and Shelf Science* **133**: 172–181.

Saintilan, N. 2009a: Biogeography of Australian saltmarsh plants. *Austral Ecology* **34**: 929–937.

Saintilan, N. 2009b: Distribution of Australian saltmarsh plants. *In* Saintilan, N. (ed): *Australian Saltmarsh Ecology.* CSIRO Publishing, Collingwood: 23–39.

Saintilan, N. & Adam, P. 2009: Preface. *In* Saintilan, N. (ed): *Australian Saltmarsh Ecology.* CSIRO Publishing, Collingwood: ix–xi.

Sánchez, J., Izco, J. & Medrano, M. 1996: Relationships between vegetation zonation and altitude in a salt-marsh system in northwest Spain. *Journal of Vegetation Science* **7**: 695–702.

Silvestri, S., Defina, A. & Marani, M. 2005: Tidal regime, salinity and salt marsh plant zonation. *Estuarine, Coastal and Shelf Science* **62**: 119–130.

Snow, A.A. & Vince, S.W. 1984: Plant zonation in an Alaskan salt marsh: II. An experimental study of the role of edaphic conditions. *Journal of Ecology* **72**: 669–684.

Sur, H. & Kukal, S. 1992: A modified hydrometer procedure for particle size analysis. *Soil Science* **153**: 1–4.

Threatened Species Scientific Committee 2013: *Subtropical and Temperate Coastal Saltmarsh Conservation Advice*. Department of Sustainability, Environment, Water, Population and Communities, Canberra: 92 pp.

Van Der Valk, A.G. & Attiwill, P.M. 1983: Above- and below-ground litter decomposition in an Australian salt marsh, *Australian Journal of Ecology* **8**: 441–447.

Vince, S.W. & Snow, A.A. 1984: Plant zonation in an Alaskan salt marsh: I. Distribution, abundance and environmental factors. *Journal of Ecology* **72**: 651–667.

Wildi, O. 2013: *Data Analysis in Vegetation Ecology*, second edition. John Wiley & Sons, Chichester: 330 pp.

Wong, V., Richardson, A.M.M. & Swain, R. 1993: *The Crustaceans and Molluscs of Tasmanian Saltmarshes*. Zoology Department, University of Tasmania: 95 pp.

(accepted 11 November 2019)

JRNL G41

Museum Victoria
71943